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ABSTRACT

GRADES OR AGES: Not specified. SUBJECT MATTER: Marine sciences. ORGANIZATION AND PHYSICAL APPEARANCE: The guide has 39 chapters, each set out in a similar pattern but with minor variations: 1) to the teacher, 2) to the student, 3) problem or purpose, 4) materials, 5) procedure, 6) questions for consideration, and 7) references. Major topics covered include salt-water aquaria, the nature of tides, use of a microscope, beach analysis, the salinity of sea water, studies of a variety of sea creatures, the analysis of marine populations, and the preparation of herbarium mounts. The guide is illustrated with drawings and some photographs. It is printed and perfect-bound with a soft cover. OBJECTIVES AND ACTIVITIES: The objectives for each lesson are given in the paragraphs on "purpose." The greater part of each chapter covers student activities. INSTRUCTIONAL MATERIALS: Equipment needed for the various activities, together with reference material, is listed in each chapter. There are separate lists of periodicals, newsletters and journals, and films. (MBM)

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THE SOURCE BOOK OF MARINE SCIENCES

1970

PREPARED AS AN N.D.E.A. SPECIAL PROJECT FOR
USE IN FLORIDA PUBLIC SCHOOLS

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DEPARTMENT OF EDUCATION
TALLAHASSEE, FLORIDA
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Many persons and organizations have provided encouragement and support to this initial effort to make greater use of local environment in the teaching of science in Florida Schools.

The members of the writing team deserve a special note of thanks for sharing their own field and laboratory experiences in this publication and to the Florida Environmental Sciences Committee for its overall direction of this effort and to its individual members for their critical review of the final draft.

Sincere appreciation is expressed for the generosity of the Florida Oceanography Society at Stuart, Florida, for making available its facilities for meetings of the Environmental Committee and providing its headquarters, complete with Marine Library, as the locale for the writing team's work in the summer of 1967, and to the Martin County School System for loan of clerical equipment for use by the writing team.

As environmental education receives increasing attention in Florida school systems, marine science is being acknowledged as uniquely effective, appropriate vehicle for learning about the total environment. The SOURCE BOOK FOR MARINE SCIENCES first distributed in 1968, is a facilitating instrument that has contributed greatly to the geometric increase of marine science programs throughout the state.

Although the original intent of the SOURCE BOOK was that of a resource material for teachers, it received such wide acclaim that the supply of copies was soon exhausted and it became inperative to reprint. Through the continuing efforts of many fine marine science educators who gave of their time, the Department of Education was able to revise and print the SOURCE BOOK. Special appreciation is entended to Mr. E. Ray Roberts, Science Department, Chairman, Martin County High School. As chairman of the Revision Committee of the SOURCE BOOK FOR MARINE SCIENCES, it was through his persistent efforts in stimulating all those concerned that the publication and distribution of this revised edition was accomplished.

INTRODUCTION

This book has been prepared in the interest of the youth of Florida experiencing more meaningful science understandings through the use of their local environment. It has been developed to assist the science teachers and public school students in making greater use of the unique environment of the "Sunshine State".

The *Source Book of Marine Sciences* is the first of an anticipated list of source books covering all environmental aspects of the state: others to be developed will cover Florida Geology, Flora and Fauna, Limnology, Meteorology, etc. The development of these additional source books is contingent on the proven worth of this, the initial effort.

The State of Florida with its exceptionally long coastline, favored by year around outdoor weather conditions, is naturally inclined toward a study of the latest scientific frontier, "Innerspace". Over the past years, with growing interest in scientific studies in the public school, an increasing number of schools began advanced studies in the marine sciences area. This prompted a meeting of science teachers, science educators, and university scientists at Tavares, Florida, to examine science program needs with respect to the seas. The examination at this meeting disclosed that the needs for the State of Florida encompassed more than just the surrounding waters but required the involvement of the total environment of our unique state. Out of the Tavares meeting the Florida Environmental Sciences Committee was formed with the task of guiding the State's public school efforts in the utilization of our local environment in collaboration with the total science program. This first effort has been centered around the marine sciences area primarily because of the great interest being shown by the young people of Florida in the potential of the seas for solutions to the social and economic problems which they must face in the next century and secondly, because of the great interest being shown by federal, state, and local governments as well as industry in the year-around potential for exploration and development of Florida waters.

The materials contained in this resource book were developed with the primary aim of assisting science teachers at all levels and subject fields to better their courses by utilization of the local environment where feasible. This book is in no way intended as a separate course of study. Where interest and conditions warrant investigation by groups of advanced students under teachers with exceptional qualification to direct such studies, this book might well serve as the backbone of a course of study to be developed on the local level.

In the overall interest of developing additional resource materials on this and other aspects of our Florida environment, teachers using these materials are encouraged to forward their reactions, recommendations and their own locally developed lab and field exercises to the Consultants for Science Education, State Department of Education, Knott Building, Tallahassee, Florida 32304.

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MARINE SCIENCE ... SYNOPSIS

TO THE TEACHER

The synopsis is about the state of the art and the potential of the sea. The material herein, when added to the locally significant marine science material, will form the basis of one or more presentations to:

1) Administrators who are interested in environmentally oriented programs.
2) Students who comtemplate taking a course in marine science.
3) Parents and parent groups.
4) Civic clubs who can offer excellent support to the program.
5) Environmentalists and other groups.

SYNOPSIS

Within one generation we have seen the development of events which have changed our concepts of the universe. I refer to the development of nuclear power, the development of rocketry which has made possible the exploration of outer space and lastly our new awareness of the potential of the sea. The sense of urgency which is now apparent in our desire to explore and exploit the sea is brought about by our economic needs due to a world wide population explosion and a rapidly growing awareness that we are destroying our own environment at a time when we should be doing everything possible to increase its productivity. It is interesting to note that the three major programs – nuclear, space and oceanic – have one thing in common, i.e., a need for quality education for those engaged in activating these programs. We do not want nuclear reactors which will work 70% of the time, we do not want space shots which are successful most of the time or submersibles which are safe most of the time. We must have as our goal success which is as nearly perfect as possible. The oceanic programs now under way and those contemplated in the immediate future can be a major factor in contributing to the welfare of all mankind and to our own national security.

Within the past 10 years there has been an explosive growth in our interest in the sea. The Science of the Sea, known as the Marine Sciences, is growing rapidly from both a military and a non-military point of view. Oceanography today is a $7 billion a year business. By 1977 it has been predicted that this figure will expand to about $24 billion. Let us examine some of the economic aspects of the sea.

GAS, OIL AND SULFUR

These can be classified as resources from under the sea. Current spending by industry in offshore activities is at a rate of over a billion dollars annually. The total investment of the free world in offshore operations is over $25 billion. At this time 16% of the world output of oil of 35.3 barrels/day is from off-shore production. Six years ago this figure was only 6%, 10 years from now 33% of the estimated world total output of 70 million bpd will be from offshore fields. The production of offshore gas has also increased at an unbelievable rate. One example of this is gas from the North Sea which may well become one of prime factors in the revival of the depressed British economy. New oil exploratory vessels with highly sophistocated equipment are now available for work at practically any ocean depth. Various types of offshore oil rigs have been developed which are supported on either mobile or fixed structures. These platforms are so large that the design and construction have become an important aspect of a new field – ocean engineering. New types of undersea pipelines and the development of super-tankers has made possible the rapid and economical transport of crude oil to the refineries and the distribution of refinery products to the ultimate consumer. Education at all levels is being encouraged in the various oil oriented disciplines.

Sulfur found in salt domes in conjunction with petroleum, is now in short supply due to its use in the rapidly expanding phosphate fertilizer industry and other chemical processes.

THE MINERAL RESOURCES OF THE SEA

From near shore, the continental shelves, the floor of the sea and the deposits from beneath the sea there are mineral deposits which are just beginning to be exploited. We have been slow in the development of methods for the economical quantitative recovery of marine minerals. Two elements which are of great industrial importance – magnesium and bromine – are now successfully separated from sea-water. The economical recovery of uranium from sea-water shows promise. More accurate shipboard instruments using magnetics, gravity, radiation, electrical resistance, conductance and inductance have made it possible to detect the presence of certain minerals. Improved seismin profiling has made it possible to examine more accurately the crust of the earth beneath the sea.

FOOD FROM THE SEA

As no accurate inventory has ever been made we are ignorant of the seafoods available in the seas of the world. The United States is now 6th place in world fish production. To revive this vital industry we need more fish census studies, new fish detection methods, better harvesting methods, wider use of newly developed processing and preservation systems and better distribution and marketing techniques. Mariculture, the artificial cultivation of marine seafood (fish, oysters, crabs, seaweed), is receiving considerable attention now and shows promise of becoming economically feasible in the near future.

The production of fish protein concentrate may be one of the answers to supplying the protein requirements of the rapidly expanding world population – particularly in the underdeveloped nations.

DRUGS FROM THE SEA

Plants and Animals of the sea with potential medicinal content are an almost unknown area of research. This could become a new and promising field of medicine. Drugs from the sea may be found which could control or destroy organisms that cause diseases, alleviate pain, stimulate or relax, increase or decrease blood clotting time, reduce blood pressure, etc.

THE ROLE OF FEDERAL AGENCIES IN THE MARINE SCIENCES

The detailed responsibilities of these groups is too extensive to discuss here. The large expenditures are here. Dept. of Defense, $256.9 million on 1968. This is concerned with oceanography as it is related to national defese administered by the Navy and the Corps of Engineers whose jurisdiction is the fresh water navigable areas of this country and coastal development and preservation. The Dept. of Interior with a 1968 budget of $73.5 million consists of agencies such as the Giological Survey, The Fed. Water Pollution Control Board, the Bureau of Commerical Fisheries, Bureau of Sport Fisheries and Wildlife, Office of Saline Water, Nat. Park Service, etc. The National Science Foundation was budgeted for $38.5 million and administers a variety of academic and basic programs among which is the new "Sea Grant" College program. The Dept. of Commerce through ESSA is concerned with weather studies and predictions. The Maritime Administration is also in the Dept. of Commerce. These are just a few of the many Federal Agencies engaged in developing the potential of the sea.

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SALT-WATER AQUARIA FOR THE LABORATORY-CLASSROOM

TO THE TEACHER

The suggestions given below constitute a resume of the authors' experiences in establishing and maintaining salt-water aquaria for classroom use. The references list contains contrary opinions, but the teacher and students are urged to read as many references as possible. In listing the various devices, the details of design are not always included. Individual needs, finances, experience, curriculum and personal choice are variables to be considered.

Most variety stores carry all-plastic filters, air stones, plastic tubing and other inexpensive aquarium accessories.

Aquaria are available from the biological supply houses. In some localities, professional aquarium supply stores construct and market their own all-glass and all-plastic aquaria.

The following topics are discussed in this section:

1. *Aquarium tanks*
2. *Salt water*
3. *Air supply*
4. *Filters and aeration*
5. *Covers*
6. *Level mark*
7. *Identification*
8. *Forceps*
9. *Dip net*
10. *Repair*
11. *Cleaning*
12. *Temperature*
13. *Light*
14. *Disease*
15. *Do's and Don'ts*

AQUARIUM TANKS

1. For general classroom use, the aquarium with a stainless steel frame, glass sides and glass bottom is recommended (Fig. 2, page 5)

   Advantages: price; shock resistance

   Disadvantages: even stainless steel will react with salt water; metal ions might affect the animals.

2. The all-glass or all-plastic aquaria are preferred by most, but are not always obtainable. No metal comes in contact with the water and the adhesives used are not toxic. A disadvantage of plastic is that it scratches easily.

3. The tanks chosen for the marine lab-classroom should include a variety of sizes: 2½ gal; 5 gal; 10 gal; and at least one or two 15 gal. For larger habitats, wooden tanks plastic painted with capacities of from 75 to 150 gal. can be constructed. (Fig. 3, page 6)

4. Wide-mouthed gallon jars of glass and/or plastic are excellent for individual student use and for series experiments with individual small animals. (Fig. 4, page 6)

5. This method of producing a series of identical small aquaria was first suggested by C. S. Perry. This "Perry Habitat" has been in continuous use in research about small fish (*Gambusia sp.*) for more than a year.

   400 ml of "pea gravel" and about 1500 ml of H_2O provide an isolated study system. There is sufficient surface absorption area for oxygen; hence, aeration probably will not be necessary.

   The original tops may be ventilated and utilized to restrain "jumpers". A single large hole may be cut and covered with plastic screening.

   The rack is constructed of 8' long strips of 1/4" or 3/8" plywood. The record bar is 1/2" plywood or a 1 x 4 board. The cusps are 6½" on center. The last cusp is 4½" from each board end. See construction detail: Rack for Perry Habitat. (See page 6)

6. A curriculum involving twenty or thirty students can be more selective in tank sizes. For daily class loads of 100 students, or more, the smaller-sized tanks enable more students to participate in individual aquarium maintenance. To estimate tank capacity in gallons, divide cubic inches by 231.

7. Aquarium tanks not fitted with feet should rest on support sticks when placed on countertops. Leaks, if any, can be discovered readily and cleaning the counter-top is facilitated.

EQUIPMENT NEEDED IF YOU PLAN TO MAKE YOUR OWN ALL GLASS AQUARIA.

General Electric, Dow-Corning or equiv. Silicone Cement Case of 12 large tubes	$34.95
Glass cutter	0.75
12 sheets alum. oxide paper (for smoothing sharp edges of glass)	6.00

½ inch masking tape – 4 rolls @.75 (To hold glass in place while cement sets)	3.00
Mineral spirits to clean glass, etc. 1 gal.	.89

Caution:

1. Do not move tank or put water in it for at least 36 hours after making it. This the time that it takes for the cement to set.
2. Be sure that the aquarium is placed on a firm level table – otherwise the bottom glass will break.
3. It is not a good policy to use any glass which has been used before. It may have scratches or unseen weaknesses. Many persons have been successful in using ¼ inch plate glass for tanks up to 50 gal. For larger tanks we strongly recommended that 7/16 inch or ½ inch plate glass be used.
4. In the final test of the tank fill only half full, then wait for several hours to fill to final level. By doing this there is less of a strain on the glass and it is easier to check for leaks.

SEA WATER FOR AQUARIUM TANKS

1. Circulating, fresh sea water is the first choice. Location of the school, of course, rarely permits this.

2. A concrete, waterproofed, underground storage tank, located adjacent to the lab, provides many schools with an adequate supply of salt water. For a high school lab, a tank with a capacity of 500-750 gallons can be installed. Carrying water from the sea to the tank can be done with a dozen or so five-gallon carboys. After two or three field trips the storage tank begins to fill. Also, there will be volunteers.

Sea water can be pumped into the lab, via P.V.C.* pipe, by either: (1) a pump located inside the building; or (2) a submersible pump. The former is recommended. In either case, the pump should be non-corrosive.

If large, 50-100 gal. aquaria are used, the plumbing can be arranged so that the water is re-circulated.

3. Obtaining sea water no longer presents a problem to the inland classroom. The largest single innovation in salt water aquaria of recent years is reliable synthetic sea water. Aquarium failures generally stemmed from the absence of the trace elements. Axelrod & Vorderwinkler (1965). The newest salts have included trace minerals down to $CuSO_4$ at .00002670%!

Formulae to prepare artificial sea water appear in:

Sverdrup et al (1942)
Straughan (1964)
Turtox Service Leaflet No. 20
and many others

The formula below is from Deitrich (1957):

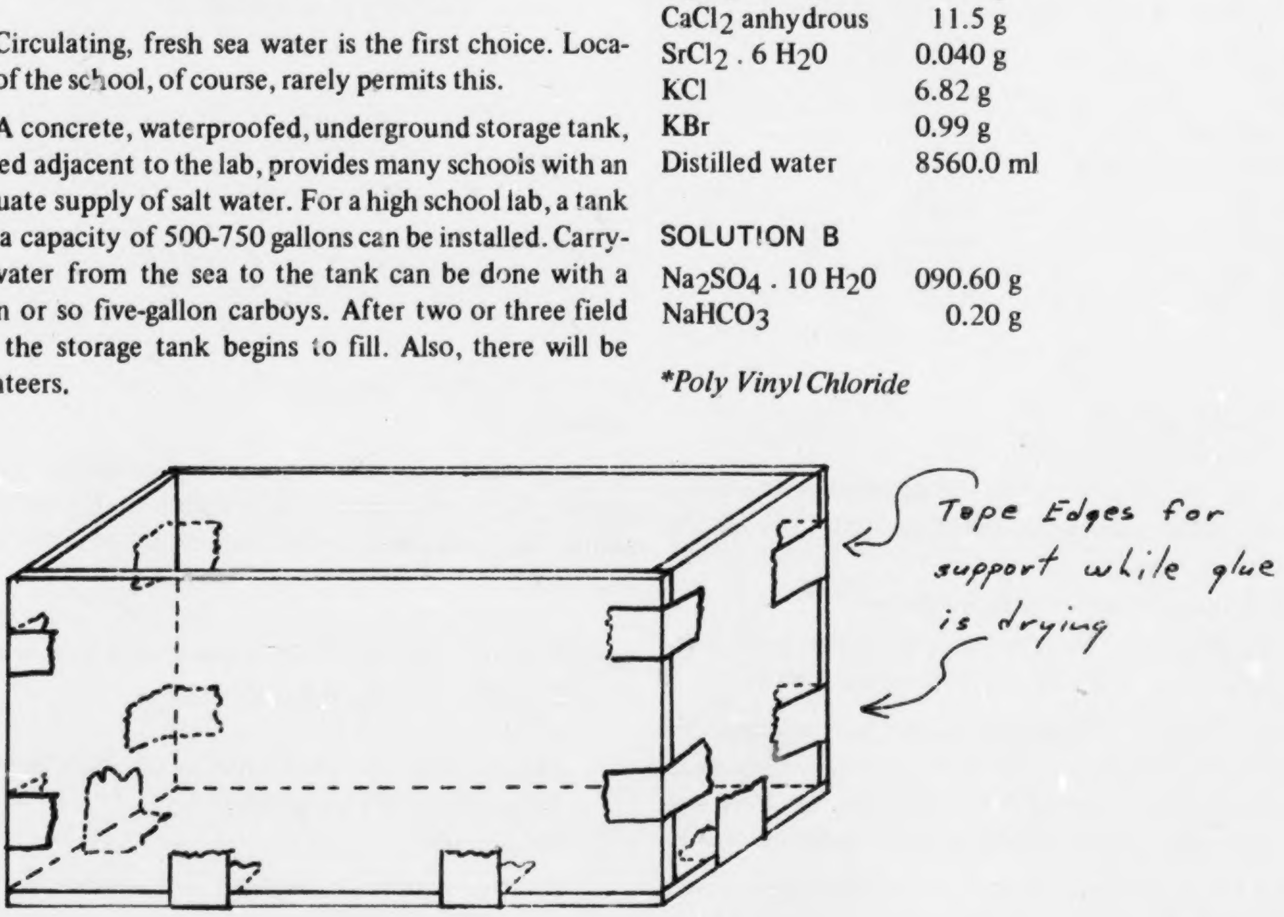
SOLUTION A

NaCl	239.0 g
$MgCl_2 \cdot 6\,H_20$	108.3 g
$CaCl_2$ anhydrous	11.5 g
$SrCl_2 \cdot 6\,H_20$	0.040 g
KCl	6.82 g
KBr	0.99 g
Distilled water	8560.0 ml

SOLUTION B

$Na_2SO_4 \cdot 10\,H_20$	090.60 g
$NaHCO_3$	0.20 g

*Poly Vinyl Chloride



NaF	0.003 g
H_3BO_3	0.027 g
Distilled Water	1000.0 ml

Solution B is added to Solution A in a thin stream while stirring.

The resulting "sea water" should stand in the dark in closed containers for more than 24 hours. Many authors recommend a week of "dark treatment".

Filter and briefly aerate. The salinity should now be equivalent to 35 parts per thousand

If several aquaria are to be established at one time, discretion being the better part of valor – buy enough commercial salts to start the year right!

Almost all biological supply houses sell the salts at reasonable prices. A few suppliers have their own brands; many represent Aquarium Systems, Inc.

Aquarium Systems, Inc., 1450 E. 289 St., Wickliffe, Ohio also sells direct. The price as of 9/10/65 for a case of salts packages varies from $12.38 to $22,80, depending on quantity purchased and the size of aquarium to be filled. The case will "salt" about 100 gallons of water: The smallest package is for a 5-gal. aquarium.

The same organization also sells several innovative devices at no small cost. They bear mentioning however: 150 gal. synthetic sea salts culture system at $2,000.00, 25 gal. culture system at $495.00, 100 gal. flowing sea water table at $1,700.00, 106 gal. mixing and storage tank at $375.00. There are other products also.

THE AIR SUPPLY

1. The peripheral air supply is ideal for the marine lab. P.V.C. pipe is installed along the backs of counters and fitted at 15" intervals with brass connecting and regulating valves. (See Fig. 2.) A central air pump furnishes air to the entire system. Some of these pumps are noisy and should be isolated from the lab, either outside or in a sound-proofed enclosure.

2. For smaller classes, individual piston pumps or vibrator pumps can service from two to eight small aquaria when fitted with gang valves.

3. For portable use, at the collecting site and for the trip back to the lab, there are small piston pumps which operate from a 6 volt dry cell battery (Example: "Air-FoBait" Corporation, P.O. Box 456, Marco Island, Florida 33937. About $13.00). This pump will service a one gallon bucket adequately. Another portable aerator, used by fishermen, utilizes a 7½ gal. plastic bucket and operates from either a 6 volt or 12 volt D.C. source.

FILTERS AND AERATORS

1. For general use, the two most popular types of filter-aerators are: (1) the sub-sand filter; and (2) the inside filter. The sub-sand type requires only a thin layer of sand or pea-gravel as a filtration device. Larger particles of detritus and uneaten food may be removed with ease. The inside filter uses either glass wool filter floss or napped nylon preferably. For classroom use washed pebbies can be substituted for charcoal. Since charcoal must be renewed frequently it becomes impractical for general use. Napped nylon is more comfortable to handle than glass wool. It can be removed easily, rinsed and put back into the filter.

2. For smaller aquaria, filter-aerators can be constructed by the students for little or no cost. For construction details see: An Inexpensive Aquarium Filter-Aerator. (Page 7)

3. Additional aeration can be provided by air stones.

AQUARIUM COVERS

It is advisable to cover the aquarium for many reasons, but the primary reason is to reduce evaporation; secondly to prevent any possible accumulation of trash.

The covers are listed below in order of preference:

plastic
glass
cellophanewrap
plywood
fiberboard

The cover should be cut to fit the outside dimensions of the tank.

THE LIQUID LEVEL MARK

As soon as the tank has been established with water, filters and animals, make a mark (use a "Dri-Mark") on the glass at the water level. When the water level drops below this line, refill with distilled water (glass-distilled). Tap-water is next best to use. The water should be at the same temperature as the water in the aquarium so that the animals will not undergo thermal shock.

IDENTIFICATION

For purposes of: (1) student responsibility; (2) student identification; and (3) animal well-being and identification, each tank must have a label. A 3x5 card suffices:

Student Name ________________ Period ____________

Date ____________________

Animals Feeding Schedule

Attach the card to the cover or to either side of the aquarium.

FORCEPS

A handy device for the student aquarist is a pair of long wooden forceps. Thin strips of wood are bound together on a wedge at one end. The tips may be left square or pointed slightly.

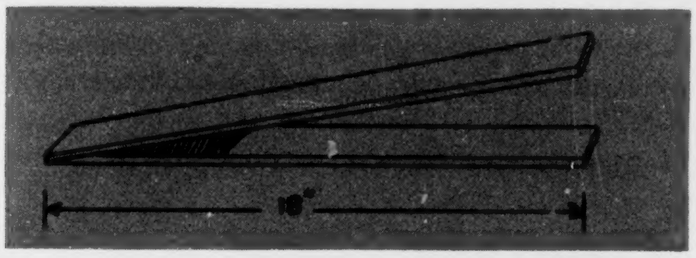


FIG. 1 *The forceps are left unpainted. These may also be purchased for about $ 1.10 each.*

DIP NET

Students may make their own dip nets or they can be purchased for the class. Due to the corrosive action of sea water, these nets will probably require replacement each year. Size for aquarium use is about 7 cm across.

REPAIR

A cracked aquarium makes a good terrarium! Corner and edge leaks can be fixed, however. Aquarium shops and biological houses have both the quick-drying plastic liquid and semi-solid cements.

CLEANING

The forceps, (See Fig. 1) dip net, and a suction type aquarium cleaner will suffice for daily detritus removal. The suction acts as a lift pump and consists of an intake tube, rubber squeeze bulb, delivery tube and filter bag. This device is available from aquarium and biological supply houses.

Portable battery-powered aquarium cleaners are also available from biological supply houses.

TEMPERATURE

The marine aquarium should be kept at the same temperature as the environment from which the specimen is taken. If this is not possible the specimens should be placed overnight in an aerated plastic bowl or dishpan. They can remain overnight in the collecting pail if aerated.

The next day those specimens that have survived and adapted to the new temperature can be introduced into the aquarium and those that are dead discarded. This method prevents having to dump an entire aquarium the day after a collecting trip because of fouling by dying and dead specimens.

If heat is necessary for the aquarium specimen, an incandescent bulb hung just outside the tank will take the place of a commerical heater.

LIGHT

The aquarium must *not* be placed so that it receives direct sunlight. Too much sunlight will cause an excessive growth of algae and will blind some species of fish. (Straughan 1964)

It is recommended that the aquarium be placed along the side of the room which receives the least amount of direct sun.

If your laboratory or classroom is air-conditioned with few or no windows, you will have to supply light through the use of fluorescent or incandescent bulbs. Do *not* use ultraviolet light bars! The possible harmful effects to your students outweigh any potential benefit. The simplest source of light is the fluorescent bar mounted above the aquarium. (Fig. 3)

The marine aquarium should receive approximately six to eight hours of light each day for best results.

DISEASE IN THE AQUARIUM

Disease in the aquarium is a constant threat but one that can be handled with an ounce of prevention.

The most common disease is "Ick". This occurs as the result of a sudden chill or a temperature change in the aquarium. The fish will become restless and scratch themselves repeatedly against sand, coral, or whatever is handy in the aquarium. A healthy specimen will occasionally scratch himself. Examine the tips of the fins for small white spots.

Treatment should begin immediately or there will be little chance of success. The treatment is poorly understood but three methods have been tried:

1. Raise the temperature of the tank to 85° or 90°F. Increase aeration and maintain temperature for four or five days.
2. Add one level teaspoonful of Sulfathiazole Sodium for each 5 gallons of water. (Straughan 1964) This is a prescription drug and must be obtained in a *water-soluble* form.
3. Add 1 drop of a 5% Methylene Blue solution per gallon of water and repeat every three days.

The treatments above apply to the following diseases by number:

salt water itch	No. 3
fungus	No. 2
parasites	No. 3
body rot	No. 2
fin and tail rot	No. 2 & 3

DO'S AND DON'TS

Do maintain constant temperature.
Do keep tank out of direct sunlight.
Do remove dead animals and uneaten food immediately.
Do maintain a constant water level. Refill with distilled or tap water.
Do keep the water well aerated.
Do keep napped nylon filters clean.
Do keep careful records.
Do keep a cover on the aquarium.
Do repair a leaking tank immediately.
Do test a tank for leaks before establishing it with sea water, animals, etc.
Do check pH periodically, "pH" paper strips will suffice.
Don't overcrowd! The rule is: one inch of fish per gallon of water. According to Axelrod and Vorderwinkler (1965), the rule is one inch of fish per two gallons of sea water.
Don't lift a full aquarium. Empty it first.
Don't overfeed! Twice a week is sufficient.

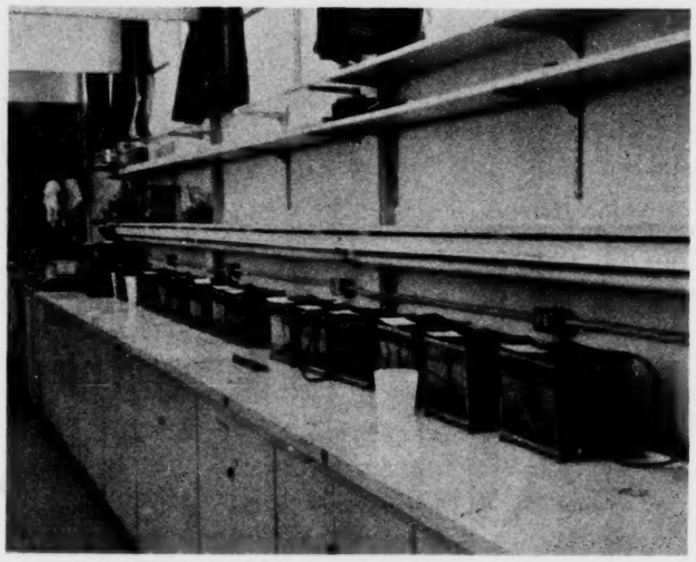


FIG. 2 *Two-gallon stainless steel and glass aquaria. Note covers and peripheral air supply.*

MATERIALS FOR AERATOR
(As Illustrated on Page 7)

1 empty plastic detergent bottle
1 piece of garden hose, 15-20 cm
1 piece of tubing long enough to reach the air supply
pebbles
napped nylon or filter floss
coat hanger wire
rubber band

OPERATION

Air bubbles and water rise in hose and percolate over the napped nylon. The water falls down through the pebbles and out of the perforations made near the neck of the plastic bottle. Clean and rinse as needed.

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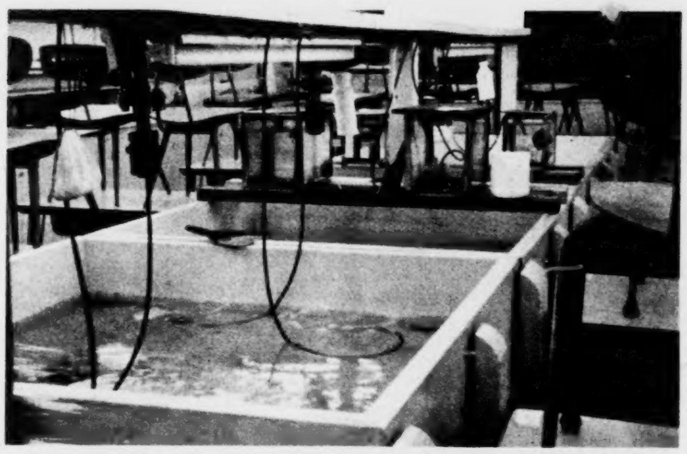


FIG. 3 | *Seventy-five gallon habitat made of wood and coated with plastic paint.*

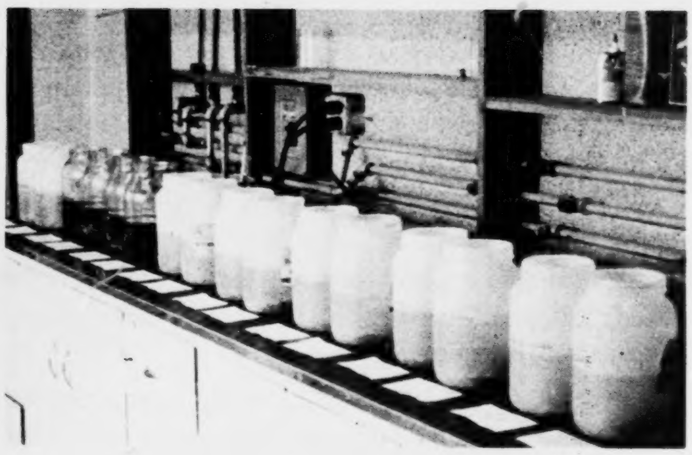
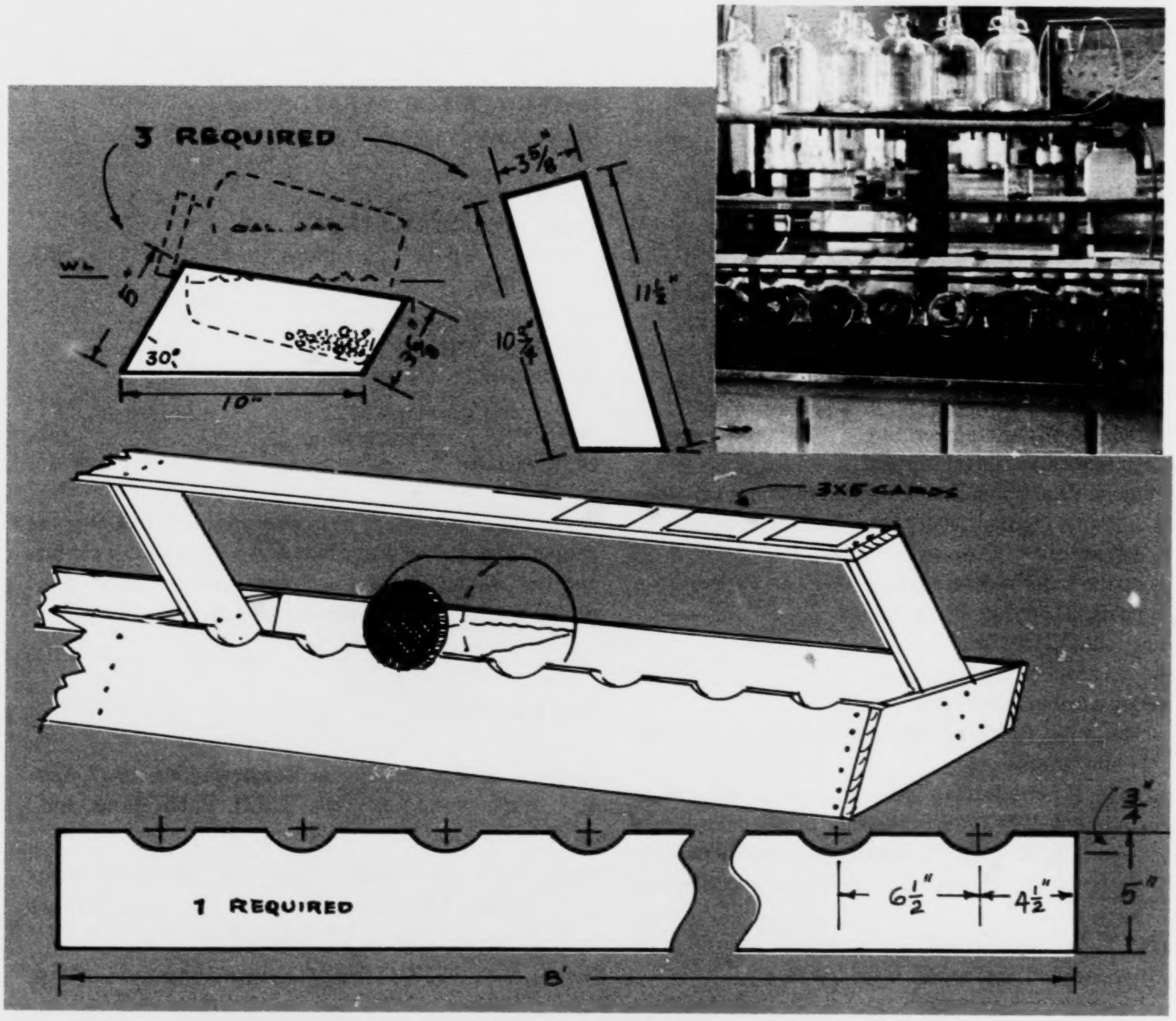
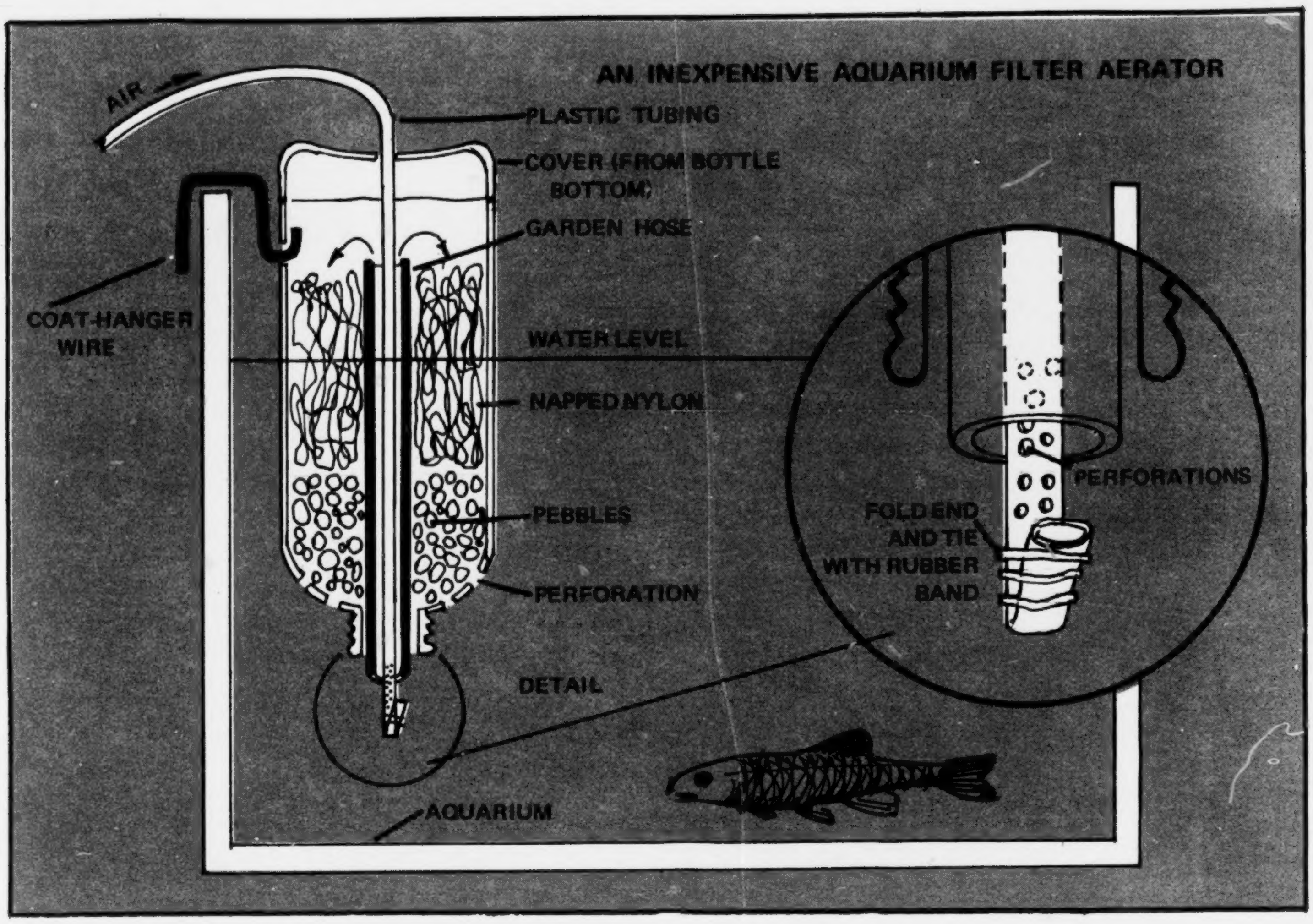


FIG. 4 | *One-gallon glass and plastic jars used as aquaria. Notice liquid level lines.*


3 REQUIRED
GAL. JAR
WL
5"
30°
10"
3 5/16"
3 5/8"
11 1/2"
10 3/4"
3X5 CARDS
1 REQUIRED
6 1/2"
4 1/2"
3/4"
5"
8'


AN INEXPENSIVE AQUARIUM FILTER AERATOR
AIR
PLASTIC TUBING
COVER (FROM BOTTLE BOTTOM)
GARDEN HOSE
COAT-HANGER WIRE
WATER LEVEL
NAPPED NYLON
PEBBLES
PERFORATION
DETAIL
AQUARIUM
PERFORATIONS
FOLD END AND TIE WITH RUBBER BAND

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USING THE 24-HOUR CLOCK

TO THE TEACHER

It is especially important that the students be thoroughly trained and practiced in the use of military time or the 24-hour time system *before* instruction in the use of the Tide Tables is given. Just as the use of the metric system should be employed, so should the 24-hour clock be used in the classroom laboratory and in the field.

Less than one laboratory period (50 min.) is required for this exercise. Discussion should be encouraged.

The exercises given here lend themselves to many variations. Answers given are for convenience (see *Conclusions for the teacher.*)

Keep in mind that standard time based upon time meridians is used, almost without exception, in published tables and reports. Therefore, local variations such as "summer time" or "daylight saving time" must be acknowledged.

The classroom clock can be converted by numbering the extra hours, 13 through 24, either to the clock face or around its outer edge. If finances permit, a bona fide 24-hour wall clock can be purchased.

TO THE STUDENT

Time is defined as a measurable period during which an action exists. The manner in which time is expressed must, then, suit the purpose for which it is intended. There are many reasons for a 24-hour clock. Accuracy, not only in recording, but also in communicating, is a primary argument *for* the 24-hour clock. The terms "AM" and "PM" are not required. For instance, 2400 hours of June 10 is the same as 0000 hours June 11. Daytime and nighttime are not significant factors. There are many places on the earth that are without the sun's light for weeks at a time. At other times there are no nights. Time in a "Sea Lab" or other underwater stations would have little use for the "AM" and "PM" designations. The day begins at midnight and runs for 24 hours.

The 24-hour clock can be adapted to Greenwich Mean Time (GMT) or Universal Time (UT) for world-wide usage. The U.S. Department of Commerce Tide Tables uses the 24-hour system exclusively, as do many navigational tables and charts.

Use this method when asked, "What time is it?" In written work, at least in the field of marine science, record all times in 24-hour designations.

PROBLEM

To learn how to express 24-hour time, and how to compute time differences.

MATERIALS

wall clock (modifications optional)
pencil (with eraser)

PROCEDURE

Exercise #1, Conversiion to 24-hour Time.

Orally, 24-hour time is expressed in "hundreds": for instance, 8:00 AM would be spoken "eight hundred hours"; 2:30 PM would be spoken "fourteen-thirty hours".

The written time uses *four* digits. Convert the following to military time:

AM-PM CLOCK	24-HOUR CLOCK
1. 7:52 AM	____________
2. 7:52 PM	____________
3. 10:00 AM	____________
4. 10:00 PM	____________
5. 3:30 PM	____________
6. 5:15 PM	____________
7. 12:00 noon	____________
8. 12:00 midnight	____________
9. 12:04 AM	____________

Exercise #2, Computing Time Differences

Keep in mind that the first two digits of a time expression represent *hours*, and that the last two digits refer to minutes. Minutes amounting to 60 or more must be converted to hours. If subtracting minutes, and "borrowing" becomes necessary, remember that when you "borrow" from the second digit you are borrowing *sixty* minutes!

	1. ADDITION		2. SUBTRACTION
(a)	0240 hrs. + 0120 hrs.	(a)	1358 hrs. - 0050 hrs.
(b)	1708 hrs. + 1000 hrs.	(b)	0911 hrs. -0119 hrs.
(c)	2250 hrs. + 0120 hrs.	(c)	0234 hrs. - 0050 hrs.
(d)	1100 hrs. + 0230 hrs.	(d)	0120 hrs. - 0300 hrs.

CONCLUSIONS FOR THE TEACHER

Answers to Exercise #1:

1. 0752 hrs.
2. 1952 hrs.
3. 1000 hrs.
4. 2200 hrs.
5. 1530 hrs.
6. 1715 hrs.
7. 1200 hrs.
8. 2400 hrs. or 0000 hrs. (next day)
9. 0004 hrs.

Answers to Exercise #2:

	1. Addition		2. Subtraction
(a)	0400 hrs.	(a)	1308
(b)	0308 hrs. (next day)	(b)	0752
(c)	0010 hrs. (next day)	(c)	0144
(d)	1330 hrs.	(d)	2220 (day before)

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Tide Tables, East Coast of North and South America, Including Greenland, 1967, Coast and Geodetic Survey, U.S. Govt. Printing Office: Washington, D.C.

THE GENERAL NATURE OF TIDES, INCLUDING INSTRUCTION IN THE USE OF THE TIDE TABLES*

TO THE TEACHER

One *Tide Tables* should be available for every two students in the class. At a cost of $30.00, a set of fifteen *Tide Tables* will provide instruction for several years. In order to learn the procedure, it is not necessary to have the latest edition of the *Tide Tables.*

Even though adequate instructions are given in the *Tide Tables*, itself, the student will find these exercises an aid to learning. It is expected that the teacher will devise additional exercises for student practice.

The exercises that follow involve: (1) finding the times of high and low tides. (2) finding the range of daily tides.

A minimum of two class days should be allotted for instruction. With these procedures available to the student, practice in the use of the *Tide Tables* can be extended through home study.

The exercises are to be done in sequence.

The lab method, "The 24-Hour Clock", should precede instruction in the use of the *Tide Tables.*

TO THE STUDENT

The tide is a giant wave – a wave thousands of miles long! Twice a day (periodically only once a day in the Gulf of Mexico) the littoral (intertidal) zone is covered with water or exposed to the sun. Typical tidal curves for Key West and Pensacola, Florida, show but a few tenths of a foot difference between high water and low water. Inspection of tidal heights in Eastport, Maine, show high tides exceeding twenty feet above mean low water!

> In the Bay of Fundy alone, some 100 billion tons of water are moved in and out with each tide. The Mississippi River, by comparison, would take nearly 140 days to move as much as the daily flow of this bay. (Smith, 1965)

Knowledge of tidal movement is important to all disciplines of marine science. Accurate predictions of tidal times and heights are as essential to the field ecologist as to the pilot of a marine research vessel. The marine biologist is aware that animals of the intertidal zone have definite tidal behavior patterns. Greater ranges between high tide and low tide mean that littoral animals must adapt to long periods of submergence and exposure. Many field trips have resulted in failure because attention was not given to tide times and/or ranges.

The average time between high tides is 12 hours and 25 minutes. The moon exerts 2¼ times more gravitational pull on the earth than the sun. Even though the sun has a mass twenty-six million times greater than that of the moon, the moon is closer to the earth. (Coker, 1954). The semi-diurnal (twice a-day) tides are primarily the function of the moon.

A lunar month is 29½ days. Within this time there will be two sets of spring tides (at new moon and full moon phases) and two sets of neap tides (at first quarter and third quarter phases). Spring tide forces are nearly three times that of the neap tides.

The effects of periodic winds, variation of atmospheric pressure, rainfall, ocean topography and currents may alter local tidal predictions. The phenomenon of the seiche (pronounced saysh) which occurs in the Gulf of Mexico, for example, is primarily responsible for interruptions in the regular semi-diurnal tide schedule. The seiche is an oscillating or sloshing back and forth of enclosed bodies of water. A daily (diurnal) tide is common to residents along the coasts of the Gulf of Mexico.

**Tide Tables. 1967. East Coast North and South America Including Greenland, U.S. Govt. Printing Office: Washington, D.C. $2.00*

The phenomenon of a tidal bore is illustrated dramatically in the Petiticodiac River at the northern end of the Bay of Fundy, New Brunswick, Canada. Tidal flow is restricted by narrow channels and tidal waters are forced to enter rivers. The bore of the Amazon River (called the Pororoca) is spectacular:

> From the banks it looks like a mile-long waterfall, up to sixteen feet high and travelling up-river with a speed of twelve knots. Its roar can be heard almost fifteen miles away. (Defant, 1958).

A tidal wave is neither a wave nor a tide. Such seismic waves, caused by earthquakes and submarine disturbances, are now called tsunamis (pronounced soo-nah'-me).

Predictions of tide times and heights are made by a combination of celestial computation and local harbor records.

"The utmost that can be expected of a tide table is that it shall be correct in calm weather and with a steady barometer." (Darwin, 1962).

The tide-gauge is set up in the quiet waters of a harbor where it is not exposed to any surface action. Heights and times are recorded graphically.

PURPOSE

The following exercises are designed to help the student learn the use of the Tide Tables: to predict times and heights of high and low water.

PROCEDURE

Do the exercises in sequence by following along with a copy of the *Tide Tables.* Work out each computation. For additional practice, pick a place and a date, at random, and compare findings with the answers found by other students in the class.

Exercise A. Find the times of high and low tides for Sarasota Bay, Florida, for October 30, 1967..

Step 1. The index to Table 2 states that Sarasota, Florida, is No. 3069. Turn to Table 2 on page 237. Find No. 3069, "Sarasota, Sarasota Bay".

Step 2. Read across to under "Differences, Time". The High water time difference reads: minus (-) 1 hour 38 minutes. The Low water time difference reads: minus (-) 0 hours 58 minutes in *St. Petersburg* (the reference station). This means that high tides at Sarasota occur 1 hour and 38 minutes *before* they do in St. Petersburg, and the lows occur 58 minutes *before* they do in St. Petersburg. Now refer to the St. Petersburg Times and Heights of High and Low Waters (Table 1). These begin on page 126 for the calendar year.

Step 3. On page 129, under October, find "30M". The "M" stands for Monday. There are two highs and two lows for the day. A glance at the heights will show that the first tide, at 0542 hrs., is a low (0.6 ft.) and that the next, at 1148 hrs., is a high (2.0 ft.). The times are figured on "Navy" time. (Refer to Exercise: Using the 24-Hour Clock).

Step 4. Set up a table like this, and solve:

	Low	High	Low	High
St. Petersburg	0542	1148	1754	2354
Correction	- 058	- 138	- 058	- 138
Sarasota Bay	0444	1010	1656	2216

Step 5. Report tide times for October 30, 1967 as follows:

Low Tide at	0444 hrs.
High Tide at	1010 hrs.
Low Tide at	1656 hrs.
High Tide at	2216 hrs.

Exercise B. Find the times of high and low tides for Sarasota Bay, Florida, for Sunday, October 15, 1967, or "15SU".

Step 1. Set up a table as follows, and solve:

	High	Low	High	Low
St. Petersburg	0042	0600	1218	1842
Correction	- 138	- 058	- 138	- 058
Sarasota Bay	2304	0502	1040	1744

Step 2. Since the High of 2304 hrs. actually occurs on October 14th, we cannot report it for October 15th. Inspection of tides for October 16th shows a tide occurring at 0048 hrs.

Step 3. Correct for Sarasota:

	High
St. Petersburg	0048
Correction	- 138
Sarasota	2310 (day before)

Step 4. Report tide times for October 15, 1967 as follows:

Low Tide at	0502 hrs.
High Tide at	1040 hrs.
Low Tide at	1744 hrs.
High Tide at	2310 hrs.

Exercise C. Find the times of highs, lows, and heights, for Boothbay Harbor, Maine, October 30, 1967.

Step 1. "Differences, Height", on page 212, states that the height of High water for Boothbay Harbor, Maine, is 0.2 ft. less than the reference station, Portland, Maine. No correction for Low tide height is required.

Step 2. Compute as follows:

	Low	
Portland	0200 hrs.	0.0 ft.
Correction	- 005	0.0
Boothbay Harbor	0155	0.0 ft.
	High	
Portland	0812 hrs.	9.8 ft.
Correction	- 006	- 0.2
Boothbay Harbor	0806 hrs.	9.6 ft.
	Low	
Portland	1424 hrs.	- 0.1 ft.
Correction	- 005	0.0
Boothbay Harbor	1419 hrs.	- 0.1 ft.
	High	
Portland	2036 hrs.	10.1 ft.
Correction	- 006	- 0.2
Boothbay Harbor	2030 hrs.	9.9 ft.

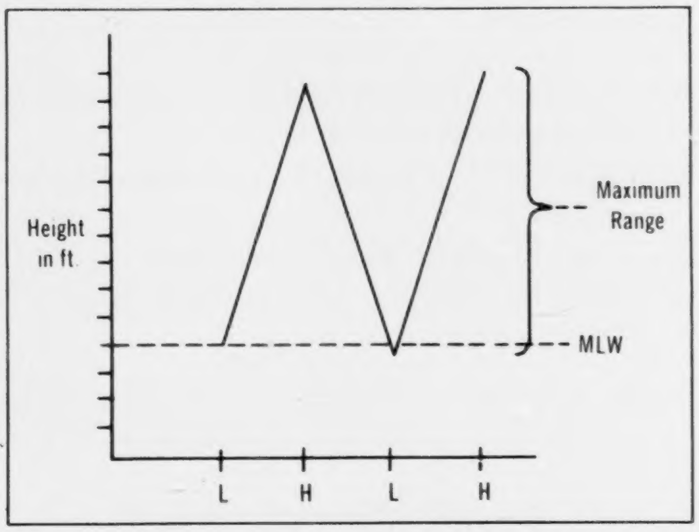
Exercise D. What is the maximum tidal range for October 30, 1967, at Boothbay Harbor, Maine?

Step 1. From Exercise D the heights of high and low waters was found to be:

Low = 0.0 ft.
High = 9.6 ft.
Low = -0.1 ft.
High = 9.9 ft.

All heights in the *Tide Table* are figured at Mean Low Water (M.L.W.).

Graphically represented:



Note: (1) that the second Low of the day is predicted to be 0.1 ft. *below* mean low water; and (2) that the second High of the day is predicted to be 9.9 ft. *above* mean low water.

Step 2. The *maximum range* for this date and this place is predicted as: *10.0 ft.*

Step 3. The *minimum range* for this date and this place is predicted as: *9.6 ft.*

QUESTIONS FOR CONSIDERATION

1. Explain why tides are different at different times and places.

2. Are there any local hazards due to tides?

3. The *Tide Tables* state "Heights are reckoned from the datum of soundings on charts of the locality which is mean low water". What significance does this have to the pilot of an ocean-going vessel?

4. Why are vertical clearances of bridges along the Intracoastal Waterway given as heights in feet above mean high water?

5. Why do the tides differ in time between Sarasota, Florida and St. Petersburg, Florida?

6. Would knowledge of tides be of any importance in a miltiary landing action?

7. Why do tidal "bulges" occur on both sides of the earth at the same time?

8. How do intertidal organisms cope with such a dynamic, ever-changing environment?

9. What are the relative positions of the earth, moon and sun during (a) spring tides, and (b) neap tides?

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CHARTING LOCAL CURRENT SYSTEMS: FIELD AND LAB EXERCISE

INTRODUCTION

In the charting of local current systems, we must first define a few terms and describe the types of currents which we might encounter near shore. We shall not consider the movements of large masses of water such as the rotation of the water of the North Atlantic Ocean (the North Atlantic gyre). The study of these local current systems is important from both a civilian and military point of view. Commercial and sport fishermen, swimmers, beach erosion experts, chart makers, and others are interested in current studies, during the daily normal changes, the changes with seasons, and the unusual changes due to storms of varying intensities. The currents and waves caused by hurricanes often change an entire coastline.

Now for a few definitions: *Tide* is the rise and fall of a large body of water, such as an ocean, due to changes in the gravitational pull of the moon and the sun on the water mass. A *tidal current* is the horizontal movement of the water. As the tide rises and falls, the currents of water move toward the shore and away from shore so we say the tidal current floods and ebbs. A *nontidal current* is any current not due to the tidal movement. *Near-shore currents* are the sum of tidal and nontidal effects and are most complicated. They are the result of many different agents acting on the water: the direction, force and duration of the wind; the rise and fall of tides; the constantly changing shape of the bottom of the near-shore water; and the sediment brought down by rivers which in turn affects bottom currents. *Estuaries* are bodies of water, salt or brackish, mostly surrounded by land, but with a connection with the sea – they also have current systems which are very difficult to study.

Many methods are in use to determine local current systems. The oldest and still the most widely-used method is the use of drift bottles. This method has been estremely valuable in measuring offshore ocean currents, but is not too satisfactory near shore. In charting nearshore and estuarine currents, dyes of various colors are dropped into the water and visual observations are made. Another indirect way of measuring bottom currents is to dye sand and determine its drift. With the increase in surfing all over the world, personal observations of local currents have been made. As waves and currents cause beach erosion, specialists in this field (Corps of Engineers, Scripps Inst. in California and others) have made valuable contributions to our knowledge of currents. Changes in the shape of the beach often give very accurate indications of current velocity and direction.

FIELD PROCEDURES FOR MEASUREMENT OF NEAR-SHORE CURRENTS

1. Use of floats outside the breaker zone – Floats can be made by students. The sketches (page 17) give some suggestions on their construction. The effect of the wind should be reduced by having as little of the float above the water as possible. Currents beyond the breaker zone are often weak, so there should be a large float surface for water reaction. Floats should be released along the entire length of the area to be studied. Each float should have an identifying flag or marker. Measurements should be made through several tidal cycles and for as long as a one-year period.

2. The use of fluorescein dye for the measurement of currents inside the breaker zone – Fluorescein is a yellowish-red dye which receives its name from the brilliant yellowish-green fluorescence of its alkaline solutions. A cup of sand with [illegible] teaspoonful of dye is wrapped in a paper towel, bound with a rubber band and tossed into the breaker zone. The direction of movement of a patch of colored water is then traced from shore, from a pier if one is nearby, or from a boat.

3. Ordinary rubber balloons filled with fresh water may also be used. They are put into the water beyond the surf zone where they float, due to the fresh water's having less density than the salt water.

QUESTIONS FOR CONSIDERATION

1. Why are currents near shore more difficult to study than currents in the deeper parts of the ocean?
2. What causes a "run-out"?
3. Why would the surface currents near shore be different from the bottom currents?
4. What is the difference between a tidal and a nontidal current?
5. What is an estuary?

CONCLUSIONS

It is easy to see that the measurement of near-shore currents is very difficult. Much work of a quantitative, statistical nature needs to be done. Every Marine Science education group should have a master chart of their own coastal area and as current studies are made, this should be put on the chart. The knowledge of currents which surfers, swimmers and fishermen gain from experience should be recorded for the benefit of everyone. For a group wishing to make a really serious study of the beaches, monthly charts should be prepared showing changes in the beach topography due to current, wave, and wind action. The study of sand transport along the beach is most interesting and could be a real contribution to the community.

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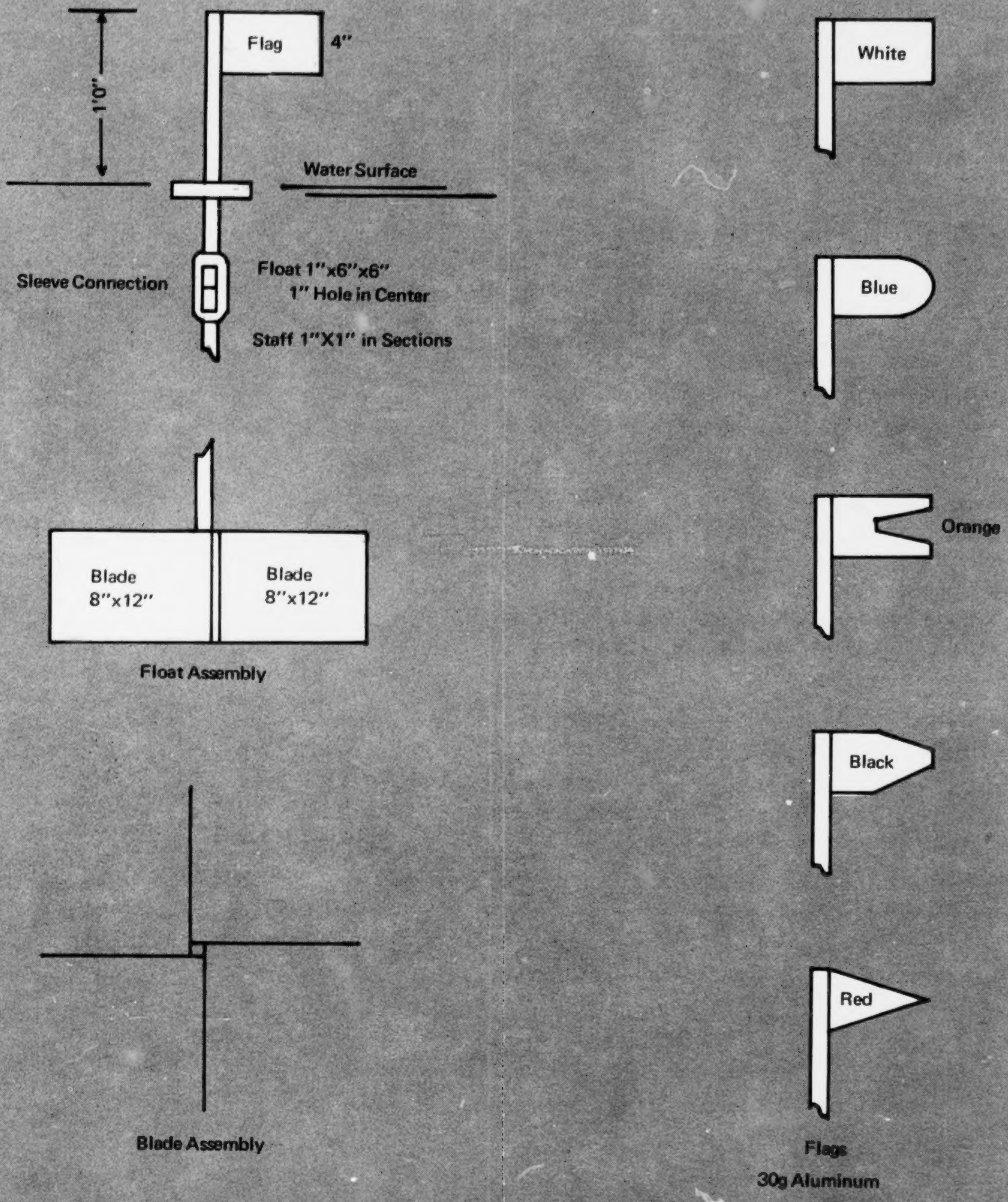

1'0"
Flag
4"
Water Surface
Sleeve Connection
Float 1"x6"x6"
1" Hole in Center
Staff 1"X1" in Sections
Blade
8"x12"
Blade
8"x12"
Float Assembly
Blade Assembly
White
Blue
Orange
Black
Red
Flags
30g Aluminum

VANE FLOAT – SUGGESTED CONSTRUCTION DETAILS

MEASURING WITH A MICROSCOPE

INTRODUCTION

From the standpoint of communication, it is essential that the student of marine science be able to view a microscopic organism and record its size. With this datum, the relative size of the organism in a drawing or photograph can be stated.

COMMENTS TO THE TEACHER

There are various types of attachments and devices for a microscope which will provide greater accuracy in measurement. Some fit beneath the ocular lens and are provided with grids and ruled lines. Many microscopes have a built-in micrometer in order to measure depth or thickness.

This particular lab procedure is given with full realization of its limitations in accuracy. It is believed, however, that the student can gain a better understanding of not only size relationships, but also he will have at his disposal a method of measurement regardless of which make of microscope he uses. Not all labs provide ocular measuring discs. If one type microscope is used throughout, the teacher may wish to compile all estimations submitted by the students and decide upon a standard figure for both the low and high power sizes.

PROBLEM UNDER INVESTIGATION

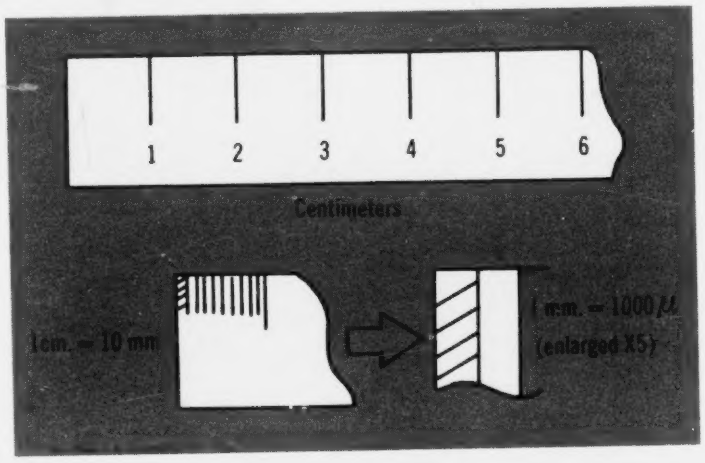
A. To determine the size of a microscopic organism.

B. To determine the relative size of a drawing or photograph of a microscopic organism.

TO THE STUDENT

Examination of any marine organism is incomplete without reference to the size of the organism. It becomes the responsibility of a student of marine science, to learn the metric system and be able to measure the length of a copepod as well as the length of a shark.

The metric system enables the biologist to measure extremely small organisms in units of less than 1 millimeter in size. For purposes of this course, organism sizes under the microscope are expressed in microns (μ). 1 micron = .001 mm. In other words, there are 1000 microns in one millimeter.



Inspect your microscope and record:

1. Ocular magnification ____________________
2. Low power objective magnification ________
3. High power objective magnification ________

Microscopes differ as to ocular lenses available and number of objective lenses on the turret. So that we have a starting point for this exercise, we will assume that the ocular is 10x, the low power is 10x and the high power is 44x. If your microscope differs from this, you may adapt by substituting your magnifications. The method is the same, regardless.

MATERIALS

Compound microscope
A plastic ruler

PROCEDURE

Part I: How to measure a microscopic organism

A. To determine the size of the microscopic field

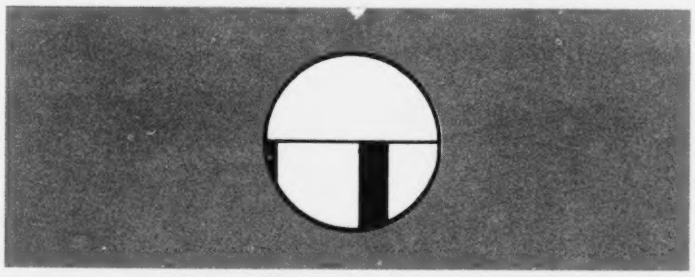
1. Measuring the low power field of vision

(a) Place the plastic ruler on the stage of the microscope so that the metric” edge covers one-half of the light-opening hole.

(b) With the low power objective in place, focus on the edge of the ruler.

(c) The *distance between* two *lines* (you will probably see no more than 2 lines) *represents one* millimeter.

(d) Move the ruler so that the middle of one line is at the edge of the field of vision:



(e) You will see that about one and one-half spaces will fit across the low power field of vision.

(f) Since 1 mm equals 1000 microns, we can estimate that the low power field of vision equals about 1500 microns (1500 μ) Note: These dimensions may vary with different microscopes.

2. Computing the high power field of vision

(a) To begin, we have the following data:

Low Power:
Ocular=10x
Objective=10
Magnification=100x (10x ocular times 10x objective)
Field diameter – 1500 microns (μ)

(b) By inspection, we find that the high power objective is 44x. Therefore, the magnification under high power is 440x (10-power ocular X 44-power objective).

(c) We also know that when we switch to high power, two things occur –

i-the size of the field is *decreased.*
ii-the amount of light coming through is insufficient and must be increased.

(d) In fact, the high power field in this case will be 4.4 times smaller than the low power field. A ratio is obtained by comparing magnifications. This is an inverse ratio, because as the *magnification* is increased, the size of the field *decreases.*

(e) We now have the following data.

High Power:
Ocular=10x
Objective=44x
Magnification=440x
Field diameter=341 microns (μ)

(f) Computation

$$\frac{100 \text{ (low power magnification)}}{440 \text{ (high power magnification)}} = \frac{1}{4.4}$$

then:

$$\frac{1}{4.4} = \frac{\text{Diameter of High Power Field}}{1500 \text{ (Diameter of Low Power Field)}}$$

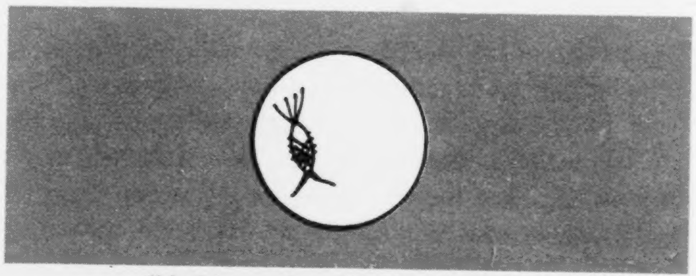
then:

$$4.4\overline{)1500\mu} = 340.9\ \mu$$

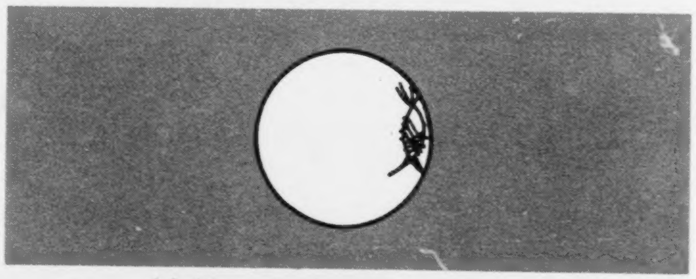
B. To determine the size of a microscopic organism

1. Estimating the size of an organism under low power

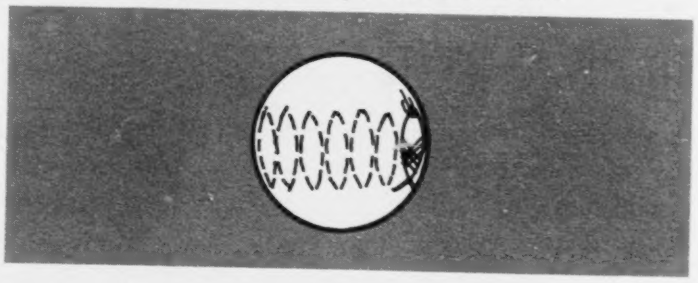
(a) Suppose you are viewing an organism under low power and wish to estimate its width, for instance:



(b) Move the slide so that the organism is oriented thus:



(c) Now, by inspection, how many of these organisms *could fit* across the field:



(d) If, for example, 7 of these could fit across the field, then the width of *each* organism would be:

$$7\overline{)1500\,\mu} = 214.5\,\mu$$

2. Estimating the size of an organism under high power

(a) Refer to parts A1 and A2. Naturally, the low power field size must be determined before proceeding here.

(b) In "Part I, A. 2. f:: we found the diameter of the high power field to be 341 μ.

(c) Again, as done in "Part I, B, 1, c" we must estimate how many organisms could fit across the diameter of the high power field of vision.

(d) This done, we divide the diameter (341 μ) by the number of organisms that will fit across the field.

(e) The answer you get will be the actual size of the organism . . . in microns.

PART II:How to Indicate the Size of an Organism on a Drawing

Note: Whether the organism is microscopic or macroscopic (large enough to observe with the naked eye), the procedure is fundamentally the same.

Step I

A. Microscopic organism

Compute the actual size of the organism from instructions in Part I.

B. Macroscopic organsim

Measure with a ruler, meter stick, calipers, etc.

Step II

Draw or photograph the organism

Step III

A. Microscope organism

Measure the drawing, in millimeters and convert to microns (1mm = 1000 μ)

B. Macroscopic organism

Measure the drawing in millimeters.

Step IV

Making sure that the *actual* measurement and the *drawing* measurement are in the same unit of measurement, i.e. both millimeters or both microns, do this:

Divide the size you got in your *drawing by* the size you got for the *actual* organism.

Step V

The answer you get will be the *size relationship* between your drawing and the actual size of the animal, the cell, or "whatever".

If your answer comes out to be 1 or more, it would be expressed:

x1, x1½, x6, x10, x250, etc.

If your answer is less than one, it is expressed as a fraction:

x½, x¼, x1/6, x1/5, etc.

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BEACH ANALYSIS

TO THE TEACHER

The newspapers are constantly featuring articles whose themes involve the disappearance of beaches. In Florida, the Miami Herald, Palm Beach Post-Times, Tampa Tribune, and Sarasota Herald-Tribune are noteworthy in this respect.

It is suggested that students might bring in current articles before the exercise is scheduled. The political and economic significance of beaches and the recreational use values should produce lively discussions.

This exercise can and should be a part of the general field trip. While some students collect biological specimens, others can collect sand, examine the sedimentation pattern and map the collection sites. If there is an extensive section in the program concerning the geologic aspects of beaches and sub-surface cores, an entire geologic field trip can be justified.

If a beach profile is made four times with the seasons, the gain or loss in beach sand is easily noted. Scientific supply houses offer the "nest" of sieves of varying sizes, composition and sieve opening. The 8" diameter brass sieves are $16-$18 each. Smaller-diametered sizes are quite adequate and are less expensive.

Part VI is best done while the overview is in progress.

TO THE STUDENT

The facts that the beaches are: 1. disappearing; 2. unavailable; and 3. crowded, are unquestioned. Knowledge of the size and types of sand particles and the factors affecting the deposition and removal of particles is important. Knowledge is always best-founded on experimentation.

This exercise has several parts. A successful exercise depends upon each getting proper consideration.

MATERIALS

magnifying glass
1 coffee can with both
    ends removed (1-
    pound size)
1 range pole

sieve set
compass
small shovel
shallow pan for drying
    samples

1 hand level (Abney)
camera (Instamatic)
hand scoops (assorted)
square of plywood to
    hold map flat
plastic bags

small paint brush with
    moderately stiff
    bristles (1/2"-1"
    wide)
rule

STATEMENT OF THE PROBLEM

To recognize, record and analyze beach characteristics. The problem is divided into six parts. They are:

Part I – A. The overview
B. The beach profile
Part II – The cross-section profile
Part III – Sampling procedure
Part IV – Moisture determination
Part V – Particle sizing
Part VI – Wave analysis

Part I

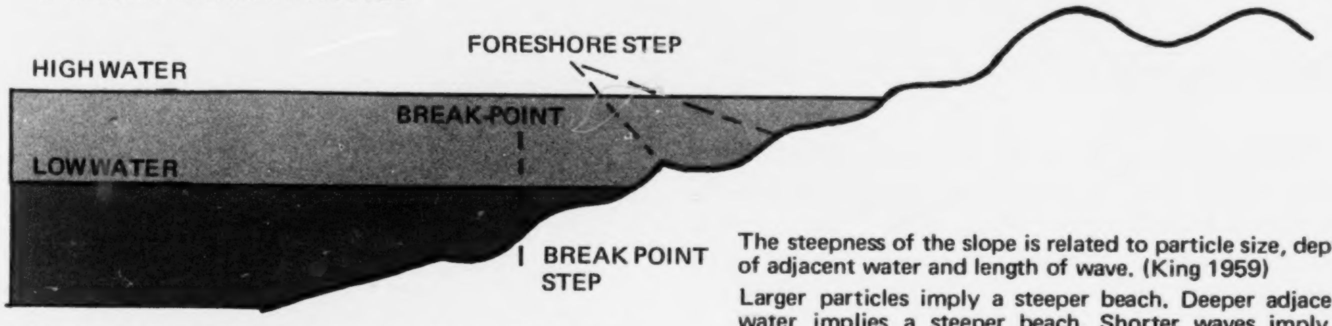
A. The overview: While standing on a promontory from which a large section of beach is visible: (1) select sampling sites; (2) map the intertidal zone; (3) map the line of vegetation; (4) circle the areas of similar vegetation. Next, plot obvious deposition characteristics such as: (1) an area of shells; (2) driftwood; or (3) drift algae. For mapping techniques refer to the attached beach characteristics sketches.

B. The beach profile: The profile-taking party should consist of three persons: a rodman, an instrument man and a recorder. Typically their duties are defined below:

The rodman holds the rangepole on designated interval points as he traverses a line perpendicular to the beach so that the instrument man can sight the elevation of the points (heights above mean low tide are positive). Rodman can either pace a regular distance between points (say 10 meters) or he can pace the metric distance between prominent

MAJOR BEACH CHARACTERISTICS TO RECOGNIZE

SHINGLE BEACH PROFILE

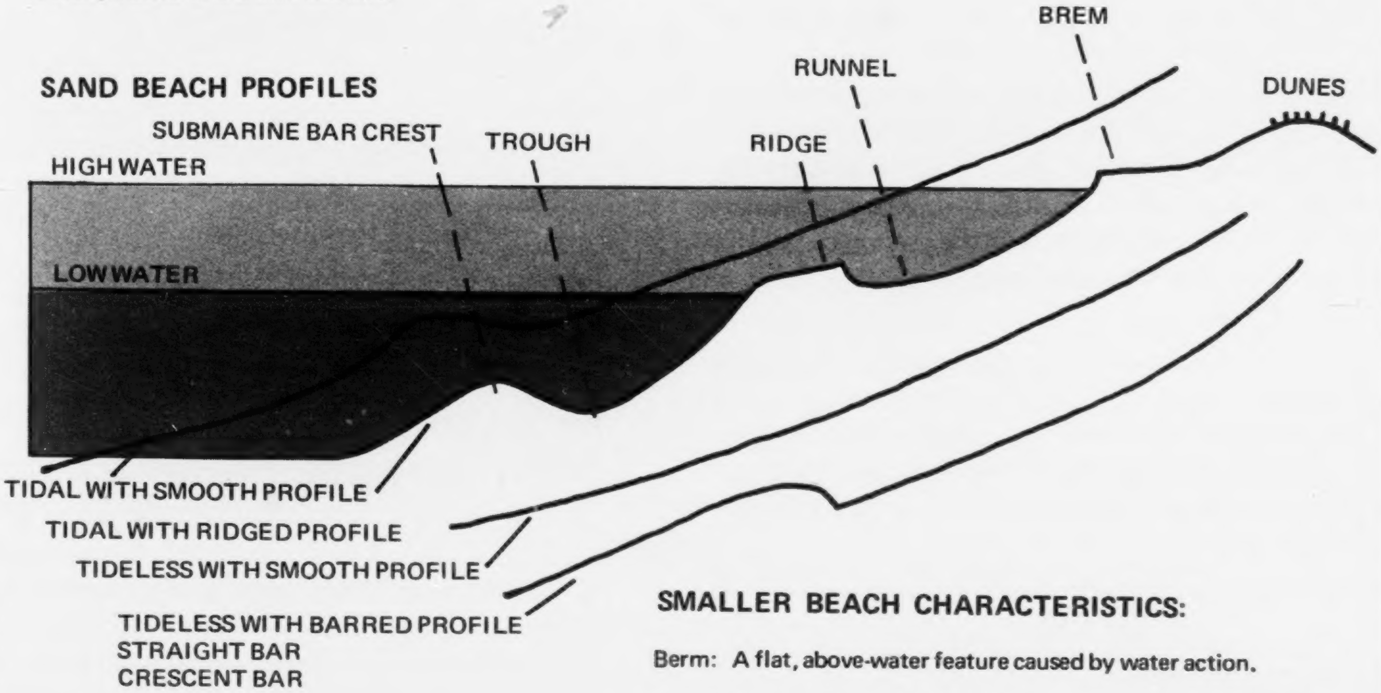

FORESHORE STEP
HIGH WATER
BREAK-POINT
LOW WATER
BREAK POINT STEP

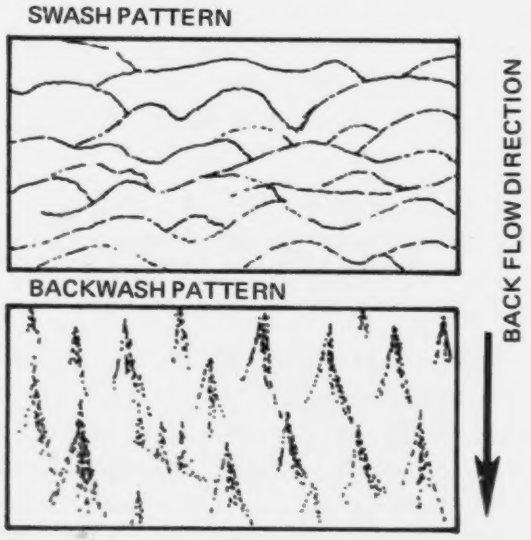
Shingle Beach: the slope is steep, similar in slope to the slope of a pile of sand poured from above. A step is usually found at the low tide line.

The steepness of the slope is related to particle size, depth of adjacent water and length of wave. (King 1959)

Larger particles imply a steeper beach. Deeper adjacent water implies a steeper beach. Shorter waves imply a steeper beach.

SAND BEACH PROFILES


BREM
RUNNEL
DUNES
SUBMARINE BAR CREST
TROUGH
RIDGE
HIGH WATER
LOW WATER
TIDAL WITH SMOOTH PROFILE
TIDAL WITH RIDGED PROFILE
TIDELESS WITH SMOOTH PROFILE
TIDELESS WITH BARRED PROFILE
STRAIGHT BAR
CRESCENT BAR


SWASH PATTERN
BACKWASH PATTERN
BACK FLOW DIRECTION

SMALLER BEACH CHARACTERISTICS:

Berm: A flat, above-water feature caused by water action.

Bar: An underwater ridge of sand usually parallel to the shoreline.

Wave Frequency: The number of waves per minute.

Wave Velocity: Speed of wave.

Wave Length: Distance between crests.

Cusp: A series of evenly spaced crescent shaped depressions below the berm.

Swash: The last rushing water from a wave is a thin sheet. The successive swash marks appear as overlapping crescents.

Backwash Pattern: When retreating water passed over minute projections downward, delta (△) shaped marks are left.

Sand domes: As water percolates through sand, entrapped air escapes, causing dome shaped bulges. The domes usually have pinholes in them.

Ripple Marks: Parallel ridges and troughs.

"breaks" in elevation. Do *NOT* change systems in mid-transit, however.

The instrument man reads the elevation while holding the instrument level. He then calls the elevation above mean low tide (+) or below mean low tide (-).

The recorder relates the map plot to the location of the range pole, records the elevation and then locates the next map plot in anticipation of next reading.

Part II The cross-section profile

A. To profile a cut, bluff, dune or other nearly vertical surfaces:

1. Clean debris from the face of the surface. Use a sweeping motion of the shovel's edge to cut a smooth, vertical plane. All strata should show in profile.
2. Place a measuring device (range pole, meter stick, or a common object of known size such as the shovel) in a true vertical line against the cleared face.
3. Photograph the strata and measurer.
4. Slect interesting strata for sampling (see Part III – B).

B. To profile a flat.

1. Excavate a hole with a width greater than the closest focusing distance of the camera. Otherwise the picture will be out of focus. If the hole shape is deltoid (Δ) less sand needs to be excavated.
2. Follow steps 2, 3, and 4 above.
3. *Refill the hole!*

Part III

A. Surface sampling: Once a site for a surface sample removal is selected and plotted, the surface debris is removed if any.

1. Push the opened coffee can "corer" flush with the ground surface.
2. Remove the earth, sand, pebbles, shells, etc. with scoops. The contents should go directly into the plastic bag. The excavation should be carefully made to the exact level of the "corer" bottom.

B. Strata sampling: (1) Once a stratum is located (see Part II), push the "corer" into the stratum until it is flush with the surface. Care must be taken to insure that the can's contents are entirely from the selected stratum. (2) Excavate, bag and tag the samples as before.

Back in the laboratory.

Part IV Moisture determination: The moisture-holding property of earth is an indicator of the amount and types of life it can support.

A. Determine the mass of the bag of sample.

B. Determine the mass of a shallow pan.

C. Spread the contents of bag onto the pan.

D. Dry the pan of sample in a household oven at 150-200°C (300-390°F). An alternate method of drying involves the use of a drying box (see Page 149) although the drying time is longer.

E. Determine the mass of pan and contents after several hours.

F. Replace in oven for one hour. Determine mass again. If the last determination is the same as (D), the sample is dry. Otherwise return to oven until successive readings are the same.

G. Mass of water: "A" + "B" – "F" = mass of H_2O (the letters refer to data from these lettered steps.)

H. Percent of moisture:

$$\frac{\text{“G”}}{\text{“F”} - \text{“B”}} \times 100 = \%H_2O$$

Part V Particle sizing: Arrange the individual sieves in a nest such that those with the largest sieve opening are at the top. Each sieve in the set has a smaller opening than the one above. Finally there is a bottom pan and a cover. A typical sieve set comprises 5 or 6 sieves. NBS (Nat'l Bureau of Standards) sizes used for selection are 5, 7, 10, 35, 80, 250, and 325.

A. 1. Scoop dried sample onto the top sieve. 2. When the pile of material indicates that the sieve is blocked put on the top; rock the nested sieves to-and-fro. Allow the bottom to "bump" on each movement. Allow the whole stack of sieves to turn in the hand after each oscillation. 3. Periodically add more material to the top sieve until all of the sample has been graded for size. If one or more sieves or the bottom pan fills with material, this can be emptied into a bag(s) or container(s). The stack of sieves can be reassembled for continued operations.

B. Sample removal and appraisal: 1. Remove cover; 2. Agitate the top sieve so as to cause the

Date ____________

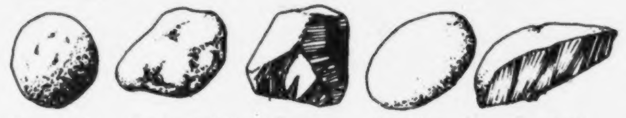
Location ____________

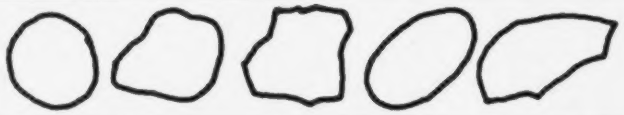
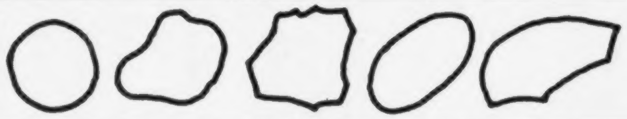
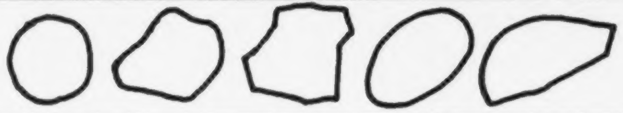
Experimenter ____________

Sample No. ____________

Moisture Content (%) ____________

Amount of dry sample ____________



A	B	C	D	E	Particle Shape Fraction	
Sieve Size	Particle Size Classification	Size Limits	Mass	$\left(\% \frac{\text{mass of fraction}}{\text{mass of sample}}\right)$	$\left(\frac{\text{Mass of shape fraction X } 100^{***}}{\text{Mass in Column D}}\right)$ percent	
Measurement by Rule	Boulder	Larger than 256 mm (10")				____ Fossils ____ Animal cast (shells)
Measurement by Rule	Cobbles	64 mm (2.5")				
7	Pebble	4mm				
10	Granule**	2mm				
230-250	Sand	1/16 mm				
See Measuring with a Microscope	Silt	1/256 mm				
Same as above	Clay	Smaller than				

** Granule as a size class may be omitted. Either "gravel" is substituted or the 2-4mm size range is included in "pebble"

*** The particle shape fraction percentage may also be found by spreading a representative amount in a shallow dish, then count and classify 100 particles. The number in each shape classification is then the percent. (see statistics) sub-sized material retained on this sieve tc pass through the openings. Stirring with the handle of the brush will help to cause these particles to pass through. 3. Empty the contents retained on the top sieve into a separate container of known mass. 4. Determine mass of container plus sample fraction. Subtract mass of the container. 5. The percent (%) is found by dividing the mass of fraction by mass of whole dry sample. Repeat steps 1-5 above for each succeeding sieve. Brush the lost particles into the container.

Part VI Wave Analysis: The frequency of waves has been noted in Part I.

A. Once this is known, count out the same number of crests. Ex. F=4/minute. Then count four crests outward.

B. Tilt hat forward or place hand above the eyes. Palm down as if to shade eyes until the 4th (example) wave is at bottom edge of hat or hand.

C. Rotate head to left or right – sight a spot on the beach on bottom edge of hat or hand.

D. Pace distance to that spot. (This is an old Boy Scout trick to measure the width of a stream.)

E. This is that distance traversed by a wave in one minute. Thus the *Velocity* of the wave is known as $\frac{\text{meters}}{\text{minute}}$.

F. Since we also know how many waves are inside the distance, a division by frequency equals *wave length.*

QUESTIONS FOR CONSIDERATION

1. Does the general appearance of a beach change with the seasons? If so, why? If not, why not?

2. Could the gain or loss of beach material be computed by comparing beach profiles from time to time?

3. Could the roundness of particles be related to the "age" of the particles?

4. Sand particles from a single stratum are:
   a. The same size and shape?
   b. Vary in size but same shape?
   c. The same size, but shape varies?
   d. Vary in size and shape?

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TURBIDITY

TO THE TEACHER

"Turbidity in water is caused by the presence of suspended matter, such as clay, silt, finely divided organic matter, plankton, and other microscopic organisms." (Standard Methods 12th Ed. 1965) Turbidity is caused by the optical property of water where light is absorbed and scattered rather than transmitted.

Of all the parameters of water analysis, turbidity determination is perhaps one of the most difficult to establish a satisfactory procedure. D.D. Jackson and G.C. Whipple compared the turbidimetric methods in 1900 in a paper printed in the MIT Quarterly. The outgrowth was that the candle turbidimeter became the laboratory standard method (Jackson Turbidimetric). Later methods of limited acceptability are photo-electric and sample comparison devices.

For field studies the Secchi disc and photometrics have been used successfully. A Secchi disc may be produced locally or purchased from suppliers.

The disc varies in diameter between 8" (20 cm) and 7½ feet (237 cm). Many commercially produced ones are about 12" (30-33 cm) in diameter. One side is white while the other side is either black or black-white marked in quadrants.

A smaller Secchi disc of about 8" diameter (22 cm) is now being marketed by the General Biological Supply House (Turtox), Chicago, Ill., and also by Gilson Slide Rule Co. of Stuart, Fla.

The Secchi disc is also used by comparing the white disc at a standard depth (1M) to a color chart. The color chart is in two parts:

The Forel Scale I-X
The Ule Scale XI-XXII

The Forel Scale is primarily for offshore blue to green waters. The yellowish to brown inshore waters match the Ule scale. These scales can be procured from Braincon Corp., Marion, Mass., and other suppliers of oceanographic instruments. They may be made from chemicals available in most high school laboratories according to the following table and directions:

> Prepare 1210 ml of solution 1; 660 ml of solution 2; 330 ml of solution 3 This is enough to prepare 4 sets of vials of 25 ml each.

Solution 1:

Measure 6.05 g of $CuSo_4.5\ H_2O$
Dissolve $CuSo_4.5\ H_2O$ in one liter of distilled H_2O
Add concentrated NH_4OH until deep blue color indicates the complete dissolving of the white precipitate. Add NH_4OH* to 1210 ml.

Solution 2:

Measure 3.3 g of $K_2CrO_4.5\ H_2O$
Measure 33 ml of NH_4OH*
Dissolve $K_2CrO_4.5\ H_2O$ in 1/2 liter of distilled H_2O.
Add NH_4OH. Add H_2O to a total volume of 660 ml.

Solution 3:

Measure 1.65 g of $CoSO_4$-7 H_2O
Dissolve $CoSO_4$ 7 H_2O in 300 ml of distilled H_2O.
Add H_2O to a total volume of 330 ml.

Make 100 ml of each Roman-numeraled mixture as directed by the table. Divide the 100 ml of mixture into four 25 ml vials. Seal each vial. Number each vial with appropriate Roman numeral. The Turtox #315A57-D is an adequate vial size and type. Compile 4 sets of the vials.

In order that the continuity of a set will not be broken, it is suggested that a wooden rack be fabricated so that the entire set of vials is visible while reading one. Place in the order of ascending Roman numerals, two racks of eleven vials to make one set. Be sure that the Secchi disc is visible while seeing each vial.

TO THE STUDENT

Turbidity is antithetical to transparency. These are physical properties of water. When light enters water from air it is reflected, refracted, absorbed, scattered and transmitted. A beam of light passing through pure H_2O has almost total transmittance. In order to detect this light, the beam needs to be pointed toward the receiver (you). The same is true for true solutions although the beam of 1 light may now appear colored. This selective absorption by color is called filtering. The colorimeter is a laboratory instrument that measures this factor. Why do underwater color photographs always seem to appear blue? Why do marine animals, who normally dwell in deep water, soon die in shallow containers?

** The NH_4OH required is made by adding 50 ml of concentrated NH_4OH to 75 ml of distilled H_2O.*

Table of Composition of each Solution in ml

Solution	I	II	III	IV	V	VI	VII	VIII	IX	X	XI
1	100	98	95	91	86	80	73	65	56	46	35
2	0	2	5	9	14	20	27	35	44	54	65
3	0	0	0	0	0	0	0	0	0	0	0
	Blue		Greenish blue		Bluish green				Green		

Solution	XII	XIII	XIV	XV	XVI	XVII	XVIII	XIX	XX	XXI	XXII
1	35	35	35	35	35	35	35	35	35	35	35
2	60	55	50	45	40	35	30	25	20	15	10
3	5	10	15	20	25	30	35	40	45	50	55
		Greenish yellow				Yellow				Brown	

To prepare the mixtures, put solution 1 in first; then 2; and then, 3; as indicated.

Both questions can have the same answer:

The infrared, red, orange and yellow light frequencies are filtered (absorbed) in the upper water levels. Thus, an animal from the "blue" deep is in a red to yellow environment. The animal may be partially blinded by the amount and color of light. His enemies now find him an easy prey.

Materials in the water that are not dissolved, but are held in suspension, produce a milky appearance. A light beam is scattered so that it can be seen from any angle. Each tiny particle reflects a minute bit of light. Since the particles are randomly oriented, the beam shows from every angle. Light that is apparent at the side angles is therefore subtracted from the light arriving at the aimed destination.

When enough suspended particles are between the source of light and the receiver of light, the object is no longer seen. If an object is lowered underwater until it can no longer be seen, the depth at which this takes place is related to the trubidity of the H_2O (vanishing point).

Such a device was developed and first used in 1865. This is the Secchi disc. Even today it is in use for measurements by oceanographers around the world.

Is there a biological significance to turbidity? An emphatic YES! Egg laying species depend upon the waters to remove CO_2 (diffusion) and to provide O_2 for absorption by the developing embryos. If these eggs are covered with a layer of silt, clay or industrial waste, the embryo development is arrested. Benthic invertebrates that feed by "pumping" water have retarded growth rates or they may die *en masse.* (See lab exercise-Pelecypod Gill – page 77). Marine plant productivity is also decreased with less light.

Turbidity caused by a high plankton count is an indicator of a favorable environment. Plankton are the foundation of the marine food chain. Ultra-clear waters are often referred to as "marine deserts" (Bahamian waters).

THE PROBLEM

A. To measure the turbidity of water.

B. To perform a color analysis of water with the Forel-Ule Scale and the Secchi disc.

MATERIAL NEEDED

Secchi disc
Forel-Ule color scale
maps
field notebook

PROCEDURE

A. To measure the trubidity of water.

1. The point of entry of the Secchi disc into H_2O should be in a shadow during a sunny day. Reflected sunlight on the surface obscures the vanishing point of the Secchi disc. There are those who can see a "sharper" vanishing point with the alternate white and black quadrants up. There are others who can do better with the white side uppermost. Try both.
2. Read and record the length of chain under water.
3. Move to next site. Repeat readings of underwater chain length (caution: The Secchi disc tends to "hydroplane" when the current is strong. Make all readings when the chain is vertical).
4. If other analysis is to be done back at the lab, a bottled sample should be taken at each Secchi-reading site.

B. To perform a color analysis with the Forel-Ule Scale and the Secchi disc.

1. Immerse the Secchi disc with the white side up. Lower the disc to a total depth of one (1) meter below the water's surface. Again, direct light reflections are to be avoided.
2. Hold the scale at arm's length so as to see both the Secchi disc and the Forel-Ule scale.
3. Next, read and record the color closest to the color of H_2O over the disc.
4. Move to next site.

STATISTICAL AND MATHEMATICAL ANALYSIS

1. Plot Secchi readings on area chart. A continuing study of this can be especially meaningful. If both chain length and color are read, a plot is made in the form of an ordered pair: (7.5, XX). This indicates that the disc "disappeared" at 7.5 meters and the water was a shade XX of brown – an unlikely occurrence. Why?
2. Connect similar readings with a continuous line similar to the contour line of a relief map.
3. Take readings at a single spot every hour during a day. Plot the hour (24-hour clock) against the depth.

QUESTIONS

1. Could a photographic light meter be used on bottled H_2O samples? How could a comparison be made with the Secchi disc field readings? (Did you ever candle an egg?)
2. Can you account for the multitudes of specimens in a shallow water biome?
3. Is there a conspicuous color change of fishes with an increase of depth? Howdo you account for this?
4. How deep does light penetrate into the ocean depths? What technique(s) can be used to verify the presence of light at a given depth?
5. In which bodies of water would you expect to find the greatest degree of turbidity? The least?

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DETERMINATION OF SUSPENDED SOLIDS IN WATER (Nonfilterable Residue)

TO THE TEACHER

To find natural water with no sediments or other solids in suspension is a rare thing. In this laboratory exercise the quantitative measure of the "nonfilterable residue", Standard Methods (1965) has implications that will later allow a nomographic analysis for dissolved carbon dioxide and other carbonate buffers.

The experimenter is introduced to gravimetrics as a general chemical procedure while making the specific test for non-dissolved materials.

The accuracy is limited by:

balance limitations
human error
drying temperature variations

Any average high school student should be able to get results that have meaning, however. A lively discussion will no doubt follow the examination of an old hot water heater's insides.

One day prior to this exercise select a package of filter discs; place in a petri dish half in the drying oven (see – "Herbarium or Instrument Drying Box" – Page 149) so that each experimenter or team can determine the mass of the filter disc. The adjusted oven temperature should be 103°-105°C.

Place as many petri dishes as there are experiments in the oven also. The tops must be ajar in order for the entrapped moisture to escape. (Hint: Place the individual filter disc in separate petri dishes at this time.)

TO THE STUDENT

Those who clean water distillation units and hot water heaters are often amazed at the residue from "clean" water. Naturally occuring water will have much more residue. The residue is normally divided into two parts. They are the filterable (dissolved) residue and the nonfilterable (suspended sediments) residue. Normal sea water has 35 grams of filterable residue in 1000 grams of water. This represents 35/1000 as a fraction. This is 35 parts per thousand (ppt). The bulk of this is Sodium Chloride, NaCl, of course. This is not the problem at hand, however.

THE PROBLEM

To determine the nonfilterable residue in a sample of water.

MATERIALS

filter apparatues – consisting of:
  vacuum filter flask, 750 ml
  one-hole stopper to fit flask #7
  filter holder, Scientific Products #F3020-2
  petri dish
  filter disc, Scientific Products #F2900-3 (GM-1)
  tweezers
  sample water
source of vacuum (aspirator)
balance
drying oven (±2°C)
large graduated cylinder

PROCEDURE

*1. Determine the mass of the dry petri dish when empty but after it has been marked for identification. (Record it)

*2. Use the tweezers to select a filter from the oven. Immediately close the cover with the filter inside.

*3. Determine the mass of filter and dish.

4. Subtract the mass of filter and dish from the mass of dish alone. This is the mass of the dry filter. (Record it.)

5. Remove filter disc and assemble filter. (If Step 1 has not been done, do so now.)

---

* *Consult the Instructor before pursuing these items.*

6. Determine the volume of sample (ml) (Record it).

7. Connect aspirator vacuum line to "arm" of filter flask. The whole filter assembly "reads" from top to bottom:

filter funnel
filter disc in holder
stopper
filter flask
vacuum line

8. Agitate the sample to keep the residue in suspension. Carefully pour this into the filter funnel.

9. When the whole sample is filtered, rinse the sample container with distilled water until it is optically clean. Filter the wash-water. If the flask overfills in the process, pour its contents into another container as directed.

10. Disassemble the filter holder. Remove the disc carefully with tweezers; place in the same petri dish as before.

11. Replace the petri dish in the oven with the cover ajar. (Be sure to keep track of this dish).

The Next Day:

12. Determine the mass of dry petri dish + filter + *nonfilterable residue.* (Record it)

13. Subtract the mass found in step 3 from step 12. *This is the mass of nonfilterable residue.* (Record it)

14. Since the volumes of samples vary, compute the amount of nonfilterable residue per liter:

$$\frac{\text{milligrams}}{\text{liter}} = \frac{\text{mass from step 13x 1000}}{\text{volume of sample in ml}} = \text{Ml}$$

GRAPHIC ANALYSIS

1. Plot graphs of: date, tide conditions, depth, and location (x-axis) against the amount of non-filterable residue per liter from the class samples. (y-axis)

2. If the class shares a single large sample, do a statistical analysis. (see "Statistical Methods")

QUESTIONS FOR CONSIDERATION

1. Does the velocity of water influence the size of the individual particles?

2. Why did the sample container have "scum" on the bottom?

*3. Look at the material on the filter under a microscope. Describe the shape and size of particles.

---

* *Consult the instructor before pursuing these items.*

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pH DETERMINATION OF SEA WATER

INTRODUCTION

Sea water is distilled water polluted by dissolved rocks, soils, minerals, and organic (plant and animal) materials. These are accumulated during the downhill rush in the creeks and rivers during that phase of the water cycle. Many of the dissolved materials then recombine through chemical reaction to form insoluble products. The remaining dissolved materials are in the form of ions, dissolved gasses and compounds.

The end product (sea water) is a mixture of solutes with the "distilled" water solvent. The final pH depends upon the excess of H^+ or OH^- ions in solution. All species of marine plants and animals have a pH tolerance. Some tolerances are "broad", i.e. pH values of 8.0 ± 1.5; and some have an extremely "narrow" pH tolerance. Fortunately, the negative ions of "weak" acids tend to resist large changes in pH. Hence, the term "buffer".

Water with large numbers of water plants tends to have higher pH values than those without. Waters having dissolved clays generally have lower values.

If the student has studied chemistry, much of the background material will be elemental. Otherwise, this exercise should extend and reinforce the acid-base relationship. It should be kept in mind that your own pH meter has specific characteristics. The instructions accompanying the instrument should be followed in preference to these.

Caution! The pH of sea water has idiosyncrasies differing from water solutions containing a single solute.

The O_2, CO_2, CO_3^{-2}, HCO_3, H^+, OH^-, the metallic ions (K^+, Na^+, Ca^{+2}) and waste product ions make this sea H_2O mixture as complex as our own body fluid. Indeed, it has often been compared to blood. Thus, to seek out one relationship between only two related ions (H^+, OH^-) is a gross simplification.

Standardized solutions must be carefully prepared and stored. Clean glassware is essential. Solutions become contaminated easily.

pH Hydrion paper covering the range of sea water is available. Try them first on saline water of known pH. Most instruments and student titrations should produce accuracies of ± 1 pH. Standard solutions prepared with saline solvents will induce a more accurate pH instrument standardization. The buffers should bracket the expected pH of 8. A single buffer at a pH of 7, 8 or 9 may be used.

INSTRUCTIONS FOR THE STUDENT

Everyone who uses the term "pH" has an idea that it has to do with degree of acidity. Not all realize that a change of one pH number is a change of the H^+ ion concentration by a factor of 10. That is: A pH of 5 has 10 times the H^+ ion concentration of pH of 6. In a like manner a pH value of 7, although it represents "neutrality", is 1000 times as acid as a pH of 10.

What is pH then –

pH is a "short hand" (lazy) way of accounting for the *available* acid ion (H^+). In a like manner there exists a *related* pOH to do the same for the available basic ion (OH).

The prevailing relationship between H^+ and OH^- is shown in two ways.

1. $H^+ + OH^- \rightarrow H_2O$ and $H_2O \rightarrow H^+ OH^-$
2. The number of H^+ times the number of OH^- ions equals a constant number at a given temperature pressure and volume of water.

   For ease of comparisons (economy of effort), chemists define the standard conditions.

   Temperature – Room temperature – 25°C
   Pressure – One atmosphere
   Volume – One liter

   Thus $[H^+]$ is a symbol representing the *amount* of H^+ ions in one liter at 25°C and at one atmosphere of pressure. You can now identify $[OH^-]$.

   The amount is also standardized in *moles* per liter. A *mole* of H^+ ions has a mass on one (1) gram. A *mole* of OH^- ions has a mass of 17 grams.

Looking again at number 2 above, write $[H^+] . [OH-] = .00000000000001 = 10^{-14}$ as the constant. Recall from algebra that $X^4 \cdot X^3 = X^7$ and $X^{-4} \cdot X^{-3} = X^{-7}$. Therefore, if the $[H^+]$ can be written as 10 raised to some power and $[OH^-]$ is done likewise, then $[H^+]\ [OH^-] = 10^{-14}$ can be done by adding the powers of ten. Since the "powers" system is designed for dilute solutions the acutal power of ten is usually negative (less than 1) and for economy of effort (Algebra: invert) we will change the sign on the power to plus.

Example $[H^+] = 1/10$ mole per liter (.1g) $= 10^{-1}$ is now called pH = 1 (the "p" is for "power":) while pOH = 13 so that pConstant = 14.

Hence pH + pOH=14.

THE PROBLEM

To find the pH of sea water.

Why is pH important? 75% of the earth's surface is best operated at a pH of 8 – 8.3. Your own blood *must* also have a similar pH. This sea-blood pH comparison has led researchers in evolution to some very interesting conclusions.

Many industrial plants discharge pollutants into the waters that lower the pH below the point where desirable organisms can live (acidify). This exercise is a very meaningful test.

MATERIALS NEEDED

indicators
test papers
clean labware
pH meter
buffers (solutions of known pH)
distilled H_2O and samples

PROCEDURE

A. Sample collecting – The size of sample will be determined by the type and number of tests to be done. The design of meter electrode will also influence the size of sample. pH meters that are available to most students will perform very well on 50 ml. of sample. Portable meters permit pH readings *in situ* (in place).

B. Operating the meter and getting the best results.

1. Standardizing the meter:
   a. Check the meter to see that it is plugged to a "hot" outlet. (For portable meters: check the battery.)
   b. Allow warm-up period (5-10 minutes).
   c. Rinse electrodes with distilled water. Wipe dry with soft, lint-free tissue.
   d. Rinse electrode with small amount of standard buffer. (Note: a marked wash bottle containing buffer from previous standardizations is handy for this. REMEMBER: *Never* pour used buffer back).
   e. Immerse electrodes in buffer solution. *Now* take the temperature of buffer. If instrument has a temperature correcting dial, set to existing temperature. Otherwise, record temperature.
   f. Activate pH "Read Control".
   g. Rotate knob marked "Adjust" or "Standardize" until meter reads the correct pH.
   h. Release pH read control.
2. Reading of pH sample:
   a. Rinse electrodes with a small amount of sample.
   b. Immerse electrodes in sample.
   c. Take temperature of sample (see 1-e above).
   d. Activate pH "Read Control".
   e. Record pH reading.
   f. Release pH "Read Control".
   g. Rinse electrodes with distilled H_2O and wipe.

C. Alternate method:

Liquid indicators and papers that show a change in color at a specific pH are available. Although there are pH Hydrion papers and others covering the expected pH of naturally occurring waters, their use is mostly confined to large variations of acidity where the decision to be made is *yes* or *no*.

Example: Is the water in the swimming pool "safe"? Yes-No. Whereas the meter is used where the specific reading is to be part of a data system for detailed analysis. Once opened, papers tend to bleach and give erratic results. Liquid indicators may cause the pH to change by interfering with the *availability* of the H^+ ions. Colorblind experimenters have difficulties here.

1. Tear off 2 inches (5 cm) of pH paper.
2. With a clean pipet place 2-3 drops of sample on one end of pH paper.
3. Visually compare the wetted end with the chart standard accompanying the pH paper.
4. Record result.
5. Repeat steps 1-4.

DATA RECORDING

Data recording for a water sample should include the following:

Sample number:
Date:
Depth:
Noted inhabitants:

Collector:
Hour: pH:
Temperature:
Climatic conditions:

QUESTIONS FOR CONSIDERATION

1. Can pH of other liquids be found?
2. If so, what body fluids can be analyzed? Blood, saliva, urine, stomach contents, and crushed plants?
3. Could one correlate pH to specimen capture?
4. If two industrial plants were upstream from a "fish-kill", how could a pH study identify the culprit?
5. Why should the electrodes be washed in the buffer and sample as recommended in the procedure?

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THE DETERMINATION OF THE SALINITY OF SEA WATER : REFRACTOMETER METHOD

TO THE TEACHER

Since many students may not be able to do quantitative analysis by "wet chemistry", a reliable yet simple instrument, the refractometer, provides the vehicle of analysis. A ray of light is bent at the interface of two media (air, solution). The degree of deviation of the light path is a function of the concentration of the solution. This deviation is *seen* – directly on the optical scale.

Since the methodology is comparatively simple, the exercise can be successfully accomplished by most students. Many students can go further toward the open end with more involved data analysis.

TO THE STUDENT

One might think of the importance of solution concentrations. The "sweetness" of tea; salty beans and the sour quality of limes are all due to the ratio of solute to solvent in a solution. Usually one can take this or leave it. Not so to the inhabitants of a liquid environment. If the sea water has too much or too little salt, inhabitant animals respond by moving into or out of certain salinities. The plants generally flourish or perish.

There are four basic methods of establishing the concentrations of solutions. Each method has advantages over the others.

1. Titration: the equipment cost is small, but the operator technique needs to be precise. The time for a determination is approximately 10-15 minutes. The chance for human error is very high. The determination must be done in the laboratory.

2. Conductance instruments: the original cost is high; the instruments often need readjustment due to rough handling and battery deterioration. Some instruments can be taken into the field. Accuracy is dependent on all the factors mentioned.

3. Optical refractometer: the refractometer has accuracy in the one part per thousand range. The instrument cost is a factor, but student error is minimized. Maintenance is nil. Data may be read with equal facility in the lab or field.

4. Hydrometry: the density of pure H_2O is one gram per milliliter, 62.4 pounds per cubic foot. The density of sea water varies with the amount of dissolved sea salts. A relationship then exists between the mass of a known volume of H_2O and the salinity of the H_2O.

Archimedes defined the fact that floating bodies displace an equal mass of liquid. That is to say, a body floats lower in a liquid of low density than in a liquid of high density.

A device that floats upright with a scale showing density of liquid at a float level is called a hydrometer. It is often found at automotive service stations. These are used to measure the percentage of anti-freeze in the radiator and in the "charge" of the battery. Although acceptable accuracy can be obtained, changes in temperature and technique cause the human error factor to limit the desirability of this technique. However, where the accuracy factor is secondary to the tracing of "gross" salinity changes, hydrometry is a standard practice.

PURPOSE

To determine the salinity of sea water.

MATERIALS

AO Goldberg Refractometer (#10402) or (#10419)
Refractive index tables (#10403)
Pipet wash bottle
Beakers
Facial tissues

PROCEDURE

1. Collecting the sample for later analysis in the laboratory.

a. *Clean* collecting bottles should be numbered. The field notes *must* locate each sample site. Be sure the sampling method is consistent. If salinity is the only analysis done, small medicine bottles or vials (1 ounce) are large enough. Samples collected for other purposes may later be analyzed without loss of accuracy providing no evaporation has occured. If the sample has partially evaporated, the original volume must be restored with distilled water.

b. Meanwhile back in the laboratory.

1. The salinometer is prepared for use by spraying the measuring prism and cover with distilled water. (Caution – The eyepiece should not get wet. Keep all liquids below the ring.)

2. Blot the distilled water with a tissue until both the cover and prism are dry. (Do *not* rub dry.)

3. Pipet 2 or 3 drops of sea water onto the prism with the cover raised and the instrument held level. (Fig. 1)

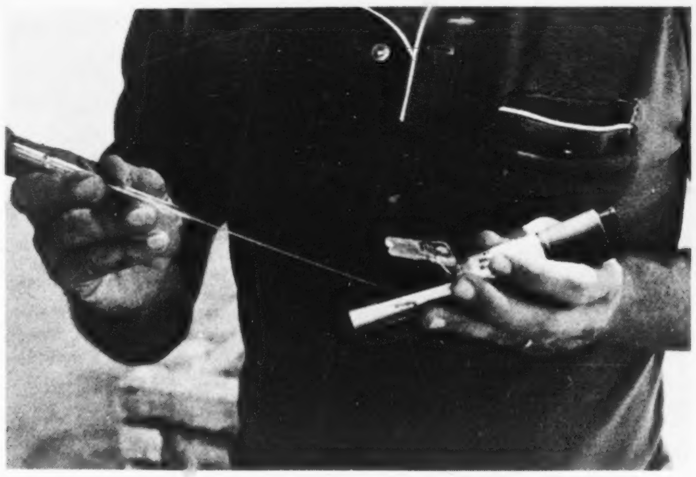

FIG. 1

4. Close cover such that the sample is squeezed into a film between cover and prism. Light finger pressure on the cover should be maintained.

5. Sight like a telescope toward a light source. The border line between the grey area above and the light yellow area below crosses scales to give the refractive index on the left. (Fig. 2)

6. The refractive index is then referred to table 47 in *Tables of Properties of Aqueous Solutions Related to Index of Refraction*. The third column headed "salinity" gives the appropriate salinity. Example: Figure 2 shows a reading of approximately 1.3340. This is the refractive index of the solution. The salinity is 5.55 parts per thousand.

2. Determination of salinity in the field requires only the salinometer, a pipet, a wash bottle and tissues. The procedure under 1.b. above could be followed. The book of tables can be read upon return to the laboratory.

3. A typical data recording should include the following:

Location (area)
Date
Climatic Conditions
Sample Number
Sample Location
Depth
Refractive Index
Salinity

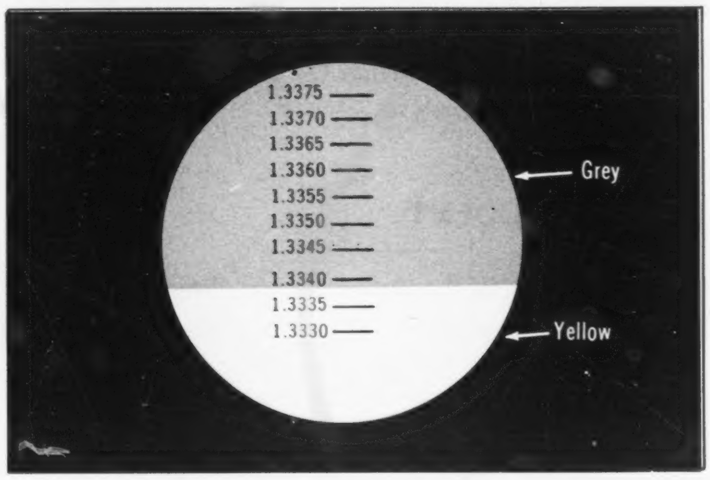

FIG. 2 | *Sight through eyepiece*

QUESTIONS

1. How can the fresh water plume from a stream be traced at sea?

2. Can one correlate the salinity tolerance of the various marine organisms?

3. How could a "Living salinometer" be compiled from the salinity preference of marine organisms?

4. Relate salinity to the density of water.

GRAPHIC ANALYSIS

Various graphs plotting salinity versus depth, plants, animals, temperature and map location. Connected areas of the same salinity will give data analysis as meaningful as contours on a relief map.

LIMITATIONS AND SOURCES OF ERROR

The accuracy of the refractive index method is largely dependent upon the knowledge or assumption that normal sea salts are the only dissolved materials in the water. Other solutes will also change the refractive index. Although the instrument is temperature corrected, extremes of temperature will induce some error.

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THE DETERMINATION OF THE SALINITY OF SEA WATER . TITRATION METHOD

TO THE TEACHER

The laboratory determination of the salinity via precipitation of the silver halides to an end point is a standard method. The "wet chemistry" process involved is titrametric. Although the experimenter may not appreciate the fine points of titration, he can easily detect the color change, measure the titrant used and correctly apply the table furnished.

The students should be made thoroughly aware of the safety problems to be encountered in any exercise wherein chemicals are employed. Silver nitrate spillage must be cleaned, rinsed and dried. Tall burets filled with titrants topple easily. The resulting mess is often wide-spread.

Overflows and spillages over the top of the buret are at eye-level or above. Eyes *must* be protected at all times (see appropriate Florida law).

Once silver nitrate is prepared, deterioration of the solution begins with exposure to light, evaporation and air borne contamination. Many chemical laboratories are "rich" in HCl, H_2S and NH_4OH (NH_3) fumes. These dissolve in the opened silver nitrate to form sediments and ionic materials. Some are interferences to good end point determinations.

The end point mechanism is that silver chloride is quantitatively precipitated before the red colored silver chromate is formed. Clumps of precipitate tend to form "refuges" for the chloride ion and the silver ion to interfere with a sharp determination. These clumps should be reduced by vigorous agitation periodically. The addition of 2 or 3 small plastic beads to the reacting vessel will be helpful to break clumps.

Although the pH of the sample is not made a part of this exercise, good results require a pH of 7-10. Adjust samples of pH below 7 with .1N NaOH.

The student must be able to read the meniscus in order to correctly do this exercise.

Preparation of the $AgNO_3$ titrant:

> Measure 27.25 grams of crystalline $AgNO_3$. Dissolve the crystals in approximately .5L of ion-free water (distilled). *Be sure all crystals have completely dissolved.* Fill with more ion-free H_2O to the one liter mark.

This solution must be stored in a dark brown bottle. If several bottles are used, the contamination of one will be a lesser problem. Each buret filling requires about 50 ml of $AgNO_3$ solution. One liter will fill 20 burets (with care).

Preparation of the K_2CrO_4 indicator:

> Add 5 grams of K_2CrO_4 yellow crystals to 100 ml of distilled H_2O.

TO THE STUDENT

Clean glassware and careful operations are essential. Begin by assuming that the table-top has been contaminated with silver nitrate by the previous class. Although $AgNO_3$ is colorless, skin will turn black in sunlight where $AgNO_3$ is present.

Dampen a paper towel with H_2O Wipe work area with the wet towel, then dry. Each should wear his apron throughout the laboratory exercise. Wear safety goggles always while in the laboratory area.

MATERIALS

1, 50 ml buret
1, 125 ml Erlenmeyer
$AgNO_3$ titrant solution
K_2CrO_4 indicator solution
*Phenophthalein
*NaOH
Distilled H_2O
2, 50 ml beakers
1, 10 ml pipet

** Optional – consult instructor before using these.*

PROCEDURE

1. Arrange the work area for maximum usability and minimum clutter. Once titration begins, nothing should interfere.

2. Fill buret with distilled H_2O. Drain buret in short spurts. Try to adjust the stopcock so as to deliver drops and a single drop on demand. Now is the time to learn the idiosyncrasies of the stopcock. Be sure it operates without leaking.

The instructor should be informed about an inoperative buret assembly immediately.

3. Pipet 10 ml of saline (sea) H_2O into the 125 ml flask. Add about 10 ml of distilled H_2O Add 2 or 3 plastic beads.

4. Put 4-6 drops of K_2CrO_4 into the flask. This is the indicator

5. Fill a 50 ml beaker with $AgNO_3$

6. Pour about 5 ml of $AgNO_3$ into the buret. Drain into the other 50 ml beaker. Pour this into sink.

7. Partially fill the buret with $AgNO_3$.Turn stopcock to fill the tip. Continue to fill the buret until there are at least 40 ml of $AgNO_3$ within the graduated scale. It is *not* necessary to fill the buret exactly to "0" or exactly to "50" (the top reading). This is time consuming.

8. Record the reading at the start (read Meniscus). The buret either has "50" or "0" or both as the top graduation. In either case record the start and end graduations. Subtract the smaller from the larger to find the volume $AgNO_3$ used in milliliters. Most burets can be read to the tenth of ml.

9. Drain the buret into the 125 ml flask of saline H_2O in short bursts (1 ml). Touch the hanging last drop in the buret with the rim of the flask so it will run into the liquid.

10. Agitate the flask. DO NOT LOSE ANY LIQUID – to make sure, use a stopper.

11. Repeat steps 9 and 10 until the first pink-orange color appears.

12. Agitate well. The clumps of precipitate must be reduced to very small particles. The flask contents should return to the original color.

13. Add $AgNO_3$ drop by drop while agitating the flask contents sufficiently to keep the precipitate particles small. When the pink color reappears, "catch" the hanging drop. Stopper. Shake vigorously. If the pink color remains, this is the end point. Otherwise, repeat Step 13.

14. Once the end point is reached calculate the volume of $AgNO_3$ used. (See 8) THIS IS THE SALINITY. However, a correction may need to be applied – consult the table below.

Salinity Corrections (Harvey, 1963)*

Salinity, S 0/00 found	Correction to be applied	Salinity, S 0/00 found	Correction to be applied
40	-0.15	22	+0.22
38	-0.08	20	+0.23
36	-0.03	18	+0.23
34	+0.03	16	+0.23
32	+0.07	14	+0.20
30	+0.11	12	+0.19
28	+0.15	10	+0.16
26	+0.17	8	+0.15
24	+0.20		

QUESTIONS

1. If an aliquot (sample fraction) with more or less than 10 ml were used (Ex – 15 ml), what additional computations must be made?

2. Assume that too much $AgNO_3$ was added to the contents of the flask. How could the experiment be "saved"?

For more questions see: "The Determination of the Salinity of Sea Water Refractometer Method".

GRAPHIC ANALYSIS

Plot various graphs of salinity versus: depth, species of plants, species of animals or temperature that were recorded where the sample was taken. Map location of connected areas of the same salinity will give data analysis as meaningful as contours on a relief map.

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MICROSCOPIC FORMS IN THE SAND : MARINE BACTERIOLOGY

TO THE TEACHER

This exercise is designed to take 3 or 4 class periods. The exercise on agar digesters and luminescent bacteria can be run concurrently by extending the time to 5 days± 1 day.

It is advisable to have the required media prepared ahead of time in tubes containing 5 ml. each, autoclaved and stored in the refrigerator until needed. If the media are stored in test tubes with approximately 5 ml. per tube, the student can melt a tube of agar and pour the required petri dish, medicine bottle, or make an agar slant. In addition, this procedure saves time when each student or team of students does not have to prepare his own media.

Make a trip to the beach and remove an area of sand at the low tide line approximately 2 cm. deep and 30 cm. square. Place the sand in a bucket and return to the laboratory.

This exercise requires excellent technique. The teacher should have familarity with general bacteriology.

TO THE STUDENT

A deserted beach appears to be a relatively sterile area, devoid of life and offering little or no protection or nutrition. If we examine the beach sand closer, we would be surprised at the abundance of life forms that exist.

Many of the life forms are microscopic bacteria, yeasts, and molds. A great many of these forms are attached to grains of sand or bits of shell. On further investigation we should realize that every time the tide changes some organisms are added and some are washed out of the beach sands.

PURPOSE

To ascertain the presence of bacteria, molds, and yeasts in beach sand.

MATERIALS

12 petri dishes or 3 oz. medicine bottles
    Bunsen burner or alcohol burner
12 slides and cover slips
    compound microscope
    crystal violet stain 2%
    sterile sea water
    inoculation loop or sterile "Q-Tips"
    grease pencil

Marine Bacteria Agar Medium

1 liter sterile sea water
10 grams peptone
15 grams agar

Marine Yeast Agar Medium

1 liter sterile sea water
20 grams agar
23 grams dextrose
1 gram Sol-U-Pro or equivalent protein
1 gram yeast extract
100 ml. Chloromycetin or Terramycin

Marine Mold Agar Medium

1 liter sterile sea water
17 grams agar
1 gram yeast extract
10 grams dextrose
100 ml. Chloromycetin, Terramycin, or penicillin

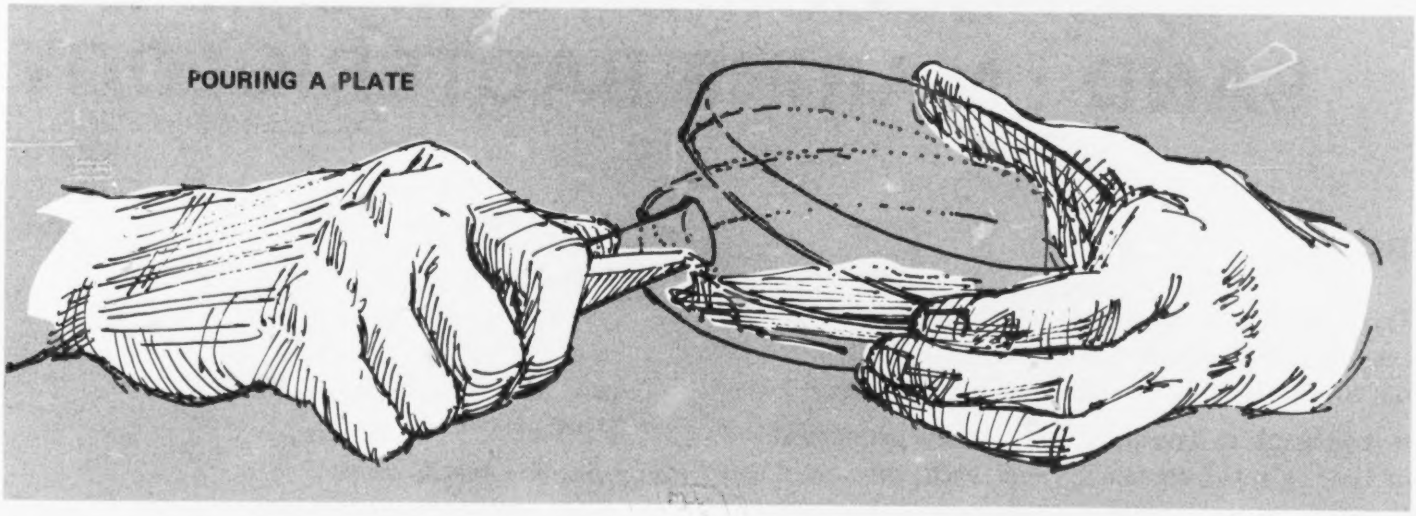
To prepare the media, the water should be brought to a simmer, then the agar and other ingredients added and stirred until they dissolve. The media should be poured into bottles and autoclaved at 15 lbs. of pressure or 120°C for 20 minutes.

PROCEDURES

1. Prepare a minimum of 3 petri dishes or medicine bottles of *each* type of medium by the method illustrated.

After melting a test tube of the desired medium, the tube should be held in the right hand with the petri dish lid opened by the left hand. The lid should be opened only enough to allow the mouth of the tube to be inserted and its contents emptied into the petri dish and then closed as rapidly as possible to prevent contamination.

The entire process should be carried out in a smooth flowing motion rather than a hurried jerking motion.


POURING A PLATE

Allow the media to harden.

2. Add 1/3 tube of the sand sample to 1/3 tube of sterile sea water, stopper, and agitate thoroughly.

3. Permit the sand to settle in the tube, then decant 1 ml. of the liquid into a sterile tube for your use.

4. Using your inoculation loop or "Q-Tip", streak two or the three petri dishes from each type of agar as illustrated (one dish from each group is to be kept as a control).

Begin at position "a" with the inoculating loop and lightly draw the loop across the agar surface in the illustrated pattern, being careful not to break the surface of the agar. On finishing at position "b" the plate should be closed immediately and the inoculation loop flamed.

5. The inoculation loop should be passed through the blue portion of a flame before and after each use.

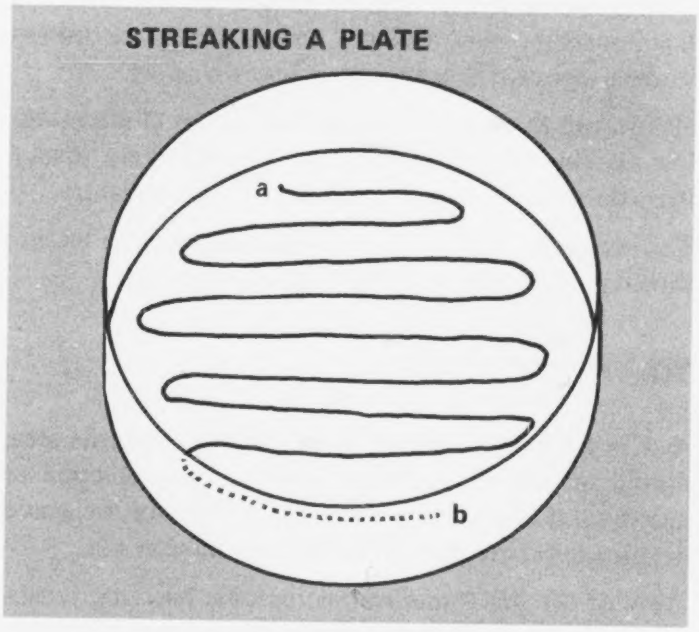
The mouth of a test tube is to be flamed before and after each opening of the tube and the tube held at a downward angle when open to prevent contamination falling into the tube from air currents.

6. Mark the streaked plates with a grease pencil to denote the type of growth expected, i.e. M = mold; Y = yeast; and B = bacteria.

7. The dishes of inoculated agar plates are to be kept in a warm, humid atmosphere for the next 4 days, preferably in an incubator with a dish of water to provide humidity.

8. The following observations are to be made each day and recorded on the chart provided:

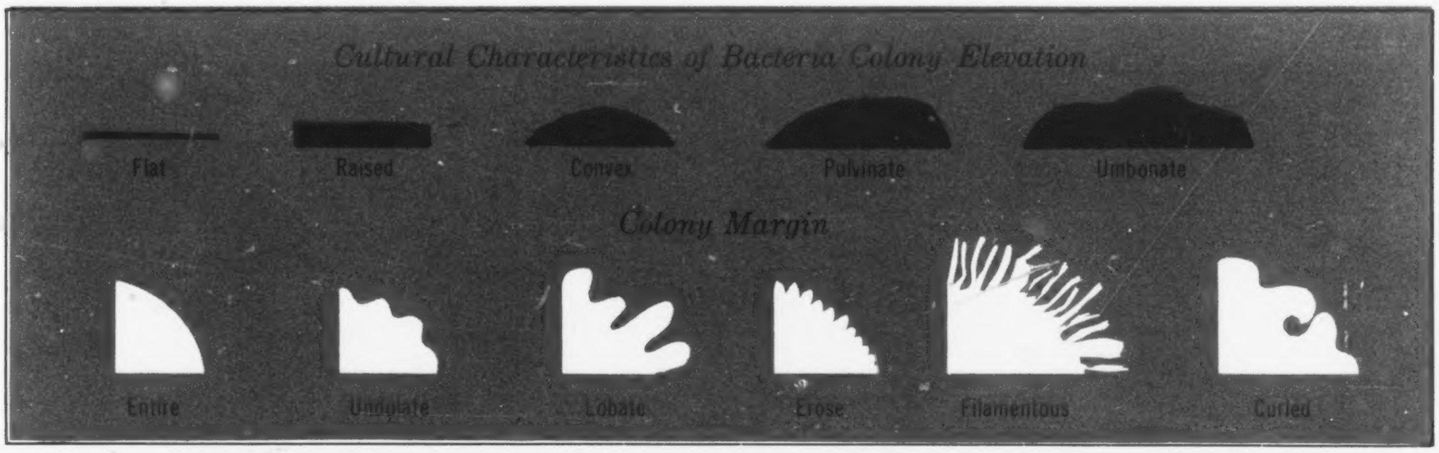
   a. number of colonies present

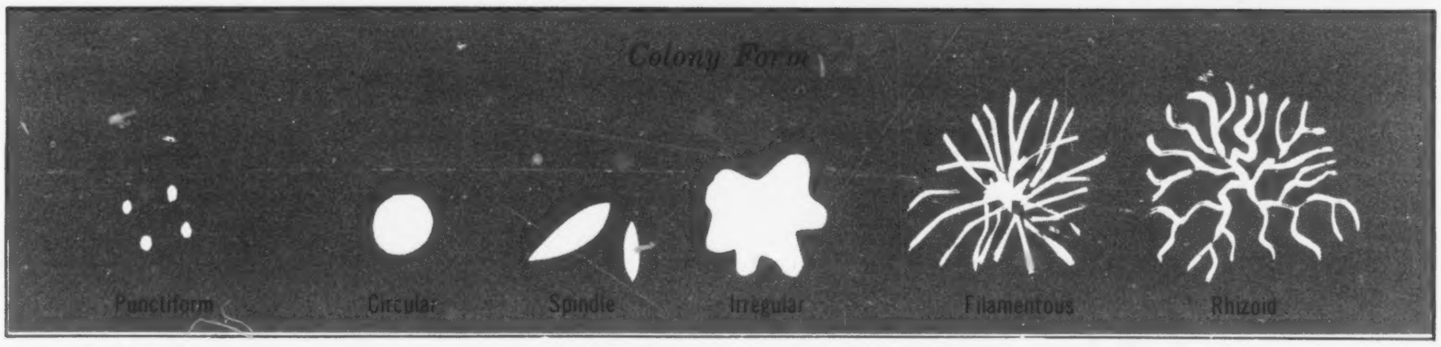

STREAKING A PLATE

   b. color, shape, and size of each colony
   c. growth pattern for each colony

From *each* type of growth on *each* plate, remove a sample of the colony with the inoculation loop and mix with a drop of sterile sea water on a clean slide. Cover with a cover glass and observe under both low and high power of the microscope.

9. After the initial observations, each slide is to be stained with crystal violet 2% stain by: (a) removing the cover slip; (b) pass the slide gently through a flame 3 or 4 times or until dry with the wet side up; (c) when the slide cools to room temperature, cover the slide with 2 or 3 drops of the stain; (d) allow the stain to remain on the slide for 20 seconds, then pour off the excess; (e) the slide should now


Cultural Characteristics of Bacteria Colony Elevation
Flat
Raised
Convex
Pulvinate
Umbonate
Colony Margin
Entire
Undulate
Lobate
Erose
Filamentous
Curled


Colony Form
Punctiform
Circular
Spindle
Irregular
Filamentous
Rhizoid

be rinsed in a beaker of clean water by immersing gently; (f) the water left on the slide is removed by blotting gently with a paper towel – DO NOT RUB.

10. The slides are now ready for observation under the microscope.

11. List and illustrate the organisms observed at this time. Identify each according to the media it was grown on.

QUESTIONS FOR CONSIDERATION

1. Did the microorganisms occur in the sea water or the sand?

2. Why would these organisms exist in the sand?

3. Why is each medium of different composition?

4. What function do these organisms have in the marine food chain?

5. At what point(s) in the procedure is contamination most likely to occur?

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Character Culture		Elevation					Margin						Form						Size				
		Flat	Raised	Convex	Pulvinate	Umbonate	Entire	Undulate	Lobate	Erose	Curled	Filamentous	Punctiform	Circular	Spindle	Irregular	Filamentous	Rhizoid	Pinpoint	Under 4-5 min.	Over 5 min.		
Bacterial Medium	No. 1																						
	No. 2																						
	No. 3																						
Yeast Medium	No. 1																						
	No. 2																						
	No. 3																						
Mold Medium	No. 1																						
	No. 2																						
	No. 3																						
Agar Medium	No. 1																						
	No. 2																						
	No. 3																						
Luminescent Medium	No. 1																						
	No. 2																						
	No. 3																						

AGAR DIGESTERS . MARINE MICROBIOLOGY

TO THE TEACHER

This exercise is designed to last 3 days or to be run concurrently with the exercise "Microscopic Forms in the Sand". In fact, it can best be run simultaneously with the above exercise.

The same instructions apply to this lab as in "Microscopic Forms in the Sand", with the exception of the source of bacteria and the media used.

Collect a series of samples of red algae (Rhodophyta) such as *Dasya* sp., *Gracilaria* sp., *Kalymenia* sp. at the beach. If at all possible, the algae should be slimy to the touch. It can be stored in the refrigerator for several days prior to its use, if it is kept enclosed in a jar or a cellophane bag.

TO THE STUDENT

In the red algae, the agar material is a carbohydrate that aids in building the plant cell wall. Since bacteria are present everywhere in the marine environment, it would be expected bacteria will be found on those algae which are used to produce agar commercially.

PURPOSE

To detect and demonstrate the presence of bacteria which use agar as food and to illustrate the variety of marine organisms which furnish support for bacterial life forms.

MATERIALS NEEDED

Red algae
3 sterile petri dishes
sterile knife
inoculating loop
test tube
iodine, 5% aqueous
sterile sea water
Agar digester medium (3 tubes)
    1 liter sterile sea water
    1 gram ammonium chloride
    0.5 gram galactose
    15 grams agar
prepare as in "Microscopic Forms in the Sand"

PROCEDURE

a. Pour the test tubes of prepared media into the sterile petri dishes and allow them to harden.

b. Select a portion of red algae with as large a surface area as possible.

c. Using a sterilized knife scrape a portion of slime from the algae surface.

d. Rinse the slime from the knife into a test tube containing 10 ml. of sterile sea water.

e. Shake the tube to disperse the slime evenly into suspension.

f. Using the inoculating loop, streak several drops of the suspension over the surface of two of the petri dishes. (The third dish becomes the control plate to demonstrate lack of contamination). Be careful not to break the surface of the agar.

g. Incubate the petri dishes at a relatively constant temperature and humidity.

ORGANIZED DATA RECORDING

Observe each petri dish daily and count the number of colonies present. Record the type of colonial growth and the occurrence of pits and depressions. Record the size change per colony on a daily basis. Check the control plate daily.

After a number of pits and depressions have developed, one of the two inoculated dishes should be flooded with iodine solution. After one minute pour off the excess iodine and record your observations. Repeat this two or three days later with the second dish and record your observations.

Analyze the iodine test on the petri dish for color production, effect on the medium by the bacteria, and other uses.

QUESTIONS FOR CONSIDERATION

1. Is the agar in depressions consumed?
2. What is the source of carbon for bacteria in the sea?
3. If bacterial growth occurs on the control plate, account for this.
4. Why is red algae selected as the source for the bacteria?

GRAPHIC ANALYSIS

Number of colonies per dish can be graphed against time required for development.

Daily size increase in colony diameter may be graphed against time.

LIMITATIONS

a. This must be carried out using sterile technique.

b. If no colonial growth is observed after 48 hours, the dish should be re-inoculated or discarded.

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BIOLUMINESCENCE

TO THE TEACHER

To obtain the required bacteria culture, a fresh marine fish is kept refrigerated for a period of two days prior to the lab exercise. This is to induce maximum bacterial growth in the outer slime covering of the fish.

This exercise requires approximately 3 days. It is suggested that this exercise be done concurrently with "Agar Digesters" and "Microorganisms in the Sand".

The luminescent bacterium, *Photobacterium fisheri*, can be obtained from a biological supply house and used rather than the fish culture.

Some organisms possess a spectacular energy-display mechanism, luminescence. "Fireflies" or "lightning bugs" are the most familiar life forms to exhibit this characteristic. It is less well-known that bacteria also possess the ability to produce light.

Bioluminescence is produced by the action of an enzyme (luciferase) on a substance known as luciferin. The reaction produces visible light. "The light emission of higher organisms may have a definite purpose . . . for attracting the sexes, as a lure for food or warning signal, perhaps for illumination . . . but it is practically impossible to assign a function of value to luminous bacteria in the above sense." (Harvey, 1952)

The smallest flash of light is seen immediately at night. This explains the numerous times a luminous wake is observed behind a boat or the wonder at the glow of the cast net used at night. This is due to the number of dinoflagellates and comb jellies that emit light when disturbed.

It has been found that bioluminescence is 100% efficient in light production. One molecule of luciferin consumed or burned produces one unit of light. This is described as "cold light" as no heat is produced. All energy is thus converted into light. (McElroy and Seliger, 1962)

Luciferin + Oxygen = Oxy-Luciferin +
Water (+light) (Russell and Yonge, 1963)

The number of organisms that exhibit bioluminescence is legion. They include bacteria, fungi, radiolarians, dinoflagellates, sponges, jellyfish, seapanseys, comb, jellies, marine worms, squids, crustaceans, clams and snails.

MATERIALS

sterile petri dishes
sterile test tubes
inoculating loop or "Q-Tip"

Marine Luminescent Bacterial Medium

- 20 grams peptone
- 10 grams glycerine
- 15 grams agar
- 1 liter sterile sea water
- Prepare according to instructions given in "Microorganisms in the Sand".

PROCEDURE

a. Prepare 3 petri dishes of medium. Use one dish as a control plate.

b. Scrape a layer of surface slime from the side of the fish, using a sterile instrument.

c. Place the scrapings in a sterile test tube containing 10 ml. of sterile sea water.

d. Agitate the test tube to mix thoroughly.

e. Use the inoculating loop or "Q-Tip" to streak two petri dishes with the solution.

f. Incubate all three petri dishes in a cool dark place (not the refrigerator) and observe daily in a dark room.

g. When luminescent colonies of bacteria are found, use the inoculating loop to scrape the colony from the surface of the agar.

h. Transfer the bacteria to a sterile test tube containing 5 ml. of sterile sea water and observe in the dark.

i. Shake the tube and repeat observations.

ORGANIZED DATA RECORDING

Observe and record daily:

- number of colonies
- size of colonies
- form of colonies
- time of appearance of each colony
- amount of luminescence per colony
- amount of luminescence in test tube before and after shaking

QUESTIONS FOR CONSIDERATION

1. Why does luminescence exist in the plant and animal world?
2. What conclusion does the refrigeration of the fish and incubation of the petri dishes lead you to make?
3. What relationship exists between the fish and bacteria?
4. How common is luminescence among the plant and animal phyla?
5. Why do some deep sea fishes possess bioluminescence?

LIMITATIONS

Time, cleanliness.

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THE TAXONOMY OF MARINE ANIMALS

A general laboratory exercise to introduce the major groups of marine animals and their characteristics.

INTRODUCTION

Taxonomy is the science of the classification of all living things. We now recognize Linnaeus' "Systema Naturae" (1758) as the beginning of an organized scientific effort to name the plants and animals of the world. Do not become discouraged with the variety of methods used to identify organisms – ideas have changed as science has progressed. Some organisms are very difficult to classify – they simply do not seem to fit into *any* scheme of classification. Remember that any modern system of taxonomy tells us much more than just the name of the animal. Its relationship to all other animals (like a family tree) and the progressive development or degeneration over millions of years can often be shown by proper classification. Remember, too, that we like to know the names of all things. Most of us are basically curious – we want to know *who* as well as *why*. If we go to a party and there are too many strange people we feel ill at ease until we are introduced. We hope to introduce you to the animals of the sea and the seashore. Since there are over 500,000 animals which have been named we can only become acquainted with a few from each major group. In science we use the binomial system of classification. For example the common blue crab is called *Callinectes sapidus* Rathbun. *Callinectes* is the genus, *sapidus,* the species, and Rathbun means that Dr. Mary J. Rathbun, who was an authority on crabs, first described this species scientifically. The word *species* means that the crabs of this group have the same characteristics and that they can mate and produce fertile offspring.

TO THE TEACHER

In the audio-visual list at the end of this exercise, we have listed the titles of a few films. We can think of no more appropriate time to use any available films related to marine animals. The diversity of marine life and many ecological principles are more beautifully illustrated by films than by any other method. The purpose of this laboratory exercise is to *introduce* the student to the major marine phyla. Detailed study of certain species will come later. This is just a wide-angle overview of the marine environment. (See the Summary of Marine Animal Phyla appended to this exercise.) The length of time spent on this lab depends on the nature of the course – whether there is only a unit on Marine Biology or an entire year. It is suggested that students start with the larger forms of the invertebrates.

TO THE STUDENT

How to collect marine specimens: At low tide specimens can simply be picked up from the beach. Collect *only* what is needed. Live specimens are always better than preserved ones. Be sure there is adequate space in the salt water aquaria at school. If there is not a salt water aquaria, preserve the specimens in 70% ethyl alcohol in sea water.

If formalin must be used remember that it will cause soft animals to disintegrate and can even damage the hard parts of many animals. If the water is not too deep for wading, nets made of hardware cloth, plastic screening, or nylon mesh can be used. Diving masks or glass-bottom buckets are a great help in locating specimens. The use of scuba by students on field trips is not recommended due to the risk involved. Every student should have a field notebook to record all details of the field trip: the date, exact location of the collecting area, weather and tide information. Information regarding wave height, water temperature, and the direction of the wind at the time of the field trip and for several days previous is helpful. Every specimen should be carefully labeled.

The Smithsonian Institution in Washington, D.C., now has over 30 million named biological specimens. With the interest in the oceans increasing so rapidly, so many specimens were being sent to the Institution that an Oceanographic Sorting Center was created. To date, over eight million specimens have been sorted and over three million specimens have been shipped to specialists for detailed study. On a small scale, the same procedure is being followed in this lab.

THE PROBLEM UNDER INVESTIGATION

The problem is to separate the animals collected on the field trip into the proper phyla. To assist in this work there is appended a summary of the Phyla of Marine Animals. It is not complete – one could spend a lifetime studying a small group of one phylum. Listed are common names of some representative animals belonging to each phylum. As common names vary considerably, the Phylum Summary should be revised to conform with indigenous common names.

MATERIALS

The materials needed and the techniques of collecting from the various marine environments are discussed elsewhere in the source book. A deep-freezer is invaluable in our taxonomic work. For example a fish can be placed on a piece of cardboard and the fins spread out and pinned. The specimen is then frozen. If glycerine is rubbed on the specimen, the frosting which obscures details can be reduced. A very large study collection can be maintained in a deep-freezer. It is particularly good for larger specimens and is ideal when laboratory time is at a premium.

PROCEDURE

The classifications of the animals will be based on their external anatomy. Under study will be the larger animals which can be seen with the unaided eye. Microscopic forms will be considered in other labs. Roughly sort the specimens according to features which are alike or dissimilar. After this rough sorting is done, see how many animals can be by phyla. Someone may have an extensive collection of seashells; another student may know the names of many species of fish. Take advantage of the human resource material first – individual knowledge. After this rough sorting, it is advisable to divide the class into groups of "specialists" – shell specialists, fish specialists, "worm" specialists, etc., who then start the classification of the specimens in detail.

ORGANIZED DATA RECORDING

Every identified specimen should be properly labeled. A suggested label form is shown below. A 3" x 5" index card should also be made for each species and maintained in a separate card file. It is hoped that in time this biological data may be correlated on a state-wide basis.

QUESTIONS FOR CONSIDERATION

1. What do we mean by the word phylum?
2. One job of a taxonomist is to name things. What other information can he give us?
3. What is a dichotomous key?
4. Most animals are classified according to their external characteristics. By what other methods could animals be classified?
5. Why is it that color is not a good basis for classification?
6. How would you define species?

Suggested Label Form

Technical name –
Common name –
Family –
Habitat –
Locale –
Description –

Date –
Collector –

A SUMMARY OF MARINE ANIMAL PHYLA

The only time that anything is simple or easy is either when we do not know anything about the subject or when someone else does the work. This is certainly true in the classification of animals. With over a million known species, it is impossible to know more than a very few – the study of one or two species may be the work of a lifetime. There are many methods of classifying animals. Used here is the general outline as suggested by Storer and Usinger in "General Zoology". Also used is Barnes "Invertebrate Zoology", "Crustaceans" by Waldo Schmitt, and Roy Miner's "Field Book of Seashore Life." Any other textbook may differ from the outline.

Phylum	Characteristics
I. Protozoa Red Tide *Noctiluca*-"night light" Foraminifera-*(Globigerina)* Radiolarians	Microscopic: single isolated cells or colonial forms. Each group has specialized way of moving. Amoeboid movement, whiplike organs or cilia.
II. Porifera Sponges	Body cylindrical, branching or irregular. Skeleton of spicules or sponging (see lab exercise on sponge spiculation). Body has many small pores and one or more large openings.
III. Coelenterata Stinging coral Portuguese man-of-war Common jellyfish Stony and other coral Sea-pens, sea pansies	Body sack-like – interior of body functions as a digestive sac. Luminescence common. All species have stinging cells (nematocysts). Live in wide variety of habitats. See lab exercise "Stinging Cells".
IV. Ctenophora Comb-jellies	Comb plates for locomotion. Only one species with nematocysts. Radially symmetrical. Usually transparent.
V. Platyhelminthes Cestodes (Tapeworms) Trematods (mostly parasitic) Flatworms Turbellaria, *Bdelloura* (found on horseshoe crab) Polyclods	Flat, soft worms without segments.
VI. Nemertinia Ribbon-worms	Flattened, elongated worms. Outer surface covered with cilia. Have proboscis to capture prey. Wide marine distribution.
VII. Rotifera Rotifers	The wheel-animals.
VIII. Nematoda Nematod worms, hairworms	Marine forms abundant; free-living and parasitic. Found in flesh of fish and in marine plants.
IX. Bryozoa Moss Animals Lamp Shells	Ancient animals. Many look like seaweeds, some resemble coral.
X. Echinodermata Starfishes Brittle stars Sea urchins Sand dollars Sea-lilies Sea cucumbers	Symmetry radial in adults; tube feet projecting ventral part of arms; body covered with delicate epidermis over mesodermal endoskeleton; which often has spines. Entirely marine.

Phylum	Characteristics
XI. Chaetognatha Arrow-worms	20-70 mm long; arrow-shaped; transparent.
XII. Mollusca Snails, bivalves (clams, oyster, scallops), chitons, octopuses, squid	About 80,000 species; most with shells, body soft.
XIII. Annelida The class Polychaeta are marine: Bristle-worms, tan-worms, etc. Earth-worms, land and fresh-water Leeches, marine, fresh-water, land some parasitic on fish	Segmented worms, setae for locomotion. There are few marine Oligochaetes (earthworms). Polychaete worms (bristle-worms) make up most of the marine forms.
XIV. Arthropoda: Crustaceans Lobsters, crabs, water fleas, copepods, barnacles.	Crustaceans vary in size from microscopic planktonic forms to lobsters 3 ft. long weighing 35 lbs. Of the approximately 80,000 species of arthropods, about 28,000 species belong to the class Crustacea. There are free-living and parasitic forms. Crustacea larva have many shapes and do not resemble adult forms. See discussion of this in labs on Crustacea.
XV. Chordata	Possess notochord
A. Acrania Tongue worms Tunicates Ascidians Lancelets	No cranium, jaws or vertebrae. These chordates are all marine, and small in size. Some forms are found in all oceans.
B. Vertebrates Cyclostomes Sharks, skates, rays Fishes-bony Birds Reptiles Mammals	Have brain enclosed in cranium. Typical body: head, neck, trunk, tail. None of this group strictly parasitic.

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AUDIO-VISUAL LIST

There are many excellent films on marine life. A few are:

The Sea – Encyclopaedia Britannica Films. (26 min. color). The infinite diversity of the structure and function of marine organism is beautifully shown.

Beach and Sea Animals – (2nd) edition Encyclopedia Films (12 min. color). Shallow water animals illustrated by some unusual underwater photography.

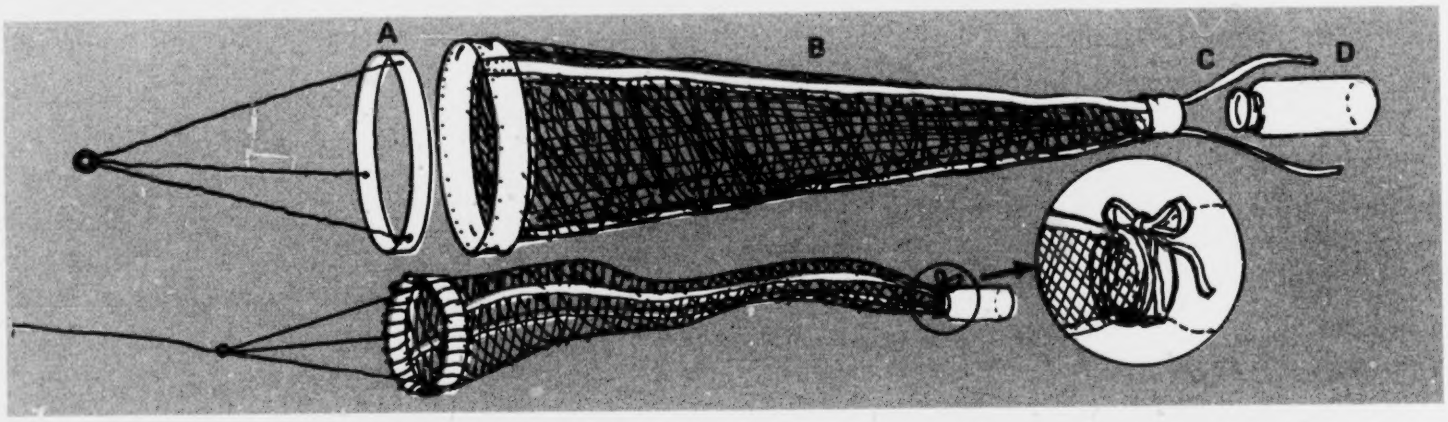
Adaptive Radiation – The Mollusks – Encyclopedia Britannica Films. (18 min. color). This covers only the mollusks.

Films on Oceanography – (1966) 3rd edition Publication C-4 National Oceangraphic Data Center, Washington, D.C. Many of the films listed in this catalog are free.

PLANKTON

TO THE TEACHER

Since a plankton net is essential for this study, instructions are included for its construction:



a. *Ring*: solidly constructed of brass, steel, etc. with diameter varying from 2 decimeters to 1 meter. Steel wire, chain or rope leaders are attached to the ring and to a single swivel at the forward end.

b. *Net*: long nylon stockings or silk bolting cloth with about 125-200 meshes to the inch are recommended. Reinforce with canvas around the openings.

c. *Ties*: sew on two canvas ribbons along the length of the net as shown. The free ends serve to secure the collecting bottle.

d. *Bottle*: sturdy plastic or glass bottle with deep threads at the neck. Size depends upon net diameter. Plastic mustard and ketchup dispensers, glass cherry, olive or babyfood jars, etc. are adequate.

Biological supply houses generally stock an assortment of plankton nets and accessories. Adapters are available to be permanently fastened into the net opening so that collecting bottles can be screwed into place rather than tied. But, best results will be achieved with the smallest mesh size, whether the net is purchased or constructed.

This particular activity centers around the collection and use of a concentrated sample which can then be diluted and distributed to an entire class. If time is available the following types of comparative samples can easily be substituted:

1. Individual samples from the same area.
2. Samples from different areas.
3. Samples taken at different times during the day (with reference to light and tide).
4. Samples taken at different depths (vertical sampling).

Pulling the net behind a slowly moving boat (1-2 knots) is one standard method of collecting plankton. For surface sampling the net should tow just below the surface. For deeper sampling tie a weight to a separate line and fasten to the front swivel. Even throwing the net from a bridge or a pier into a fast tidal current often produces excellent results.

The sample needed for this laboratory should be taken in the early morning before school . . . to be used the same day. This plan will almost insure students the rare opportunity to observe living plankton. Place sample in a larger jar or bucket and aerate gently. Dilute with a known quantity of filtered sea water. When ready to begin the lab, divide the sample into equal portions. Make available the following information to students:

Example

a. date and general location of sampling – 10/21, N. Tampa Bay
b. time of day – 0600 hrs.
c. wave activity and temperature (optional) – light chop 24°C
d. tide – low
e. depth of sample – surface
f. tow time (15 minutes is adequate for a good sample) – 15 minutes
g. mesh size – 125/inch
h. diameter of net opening (in meters) – .2

i. estimated distance net towed (in meters) – 1500

j. number of ml of team sample – 1

k. dilution – 2:1

Three periods are needed to complete all phases of the exercise. Each day includes a separate purpose, set of procedures and observation guides.

First Day: *Observation of living plankton*: student should become aware of (a) dominant forms, (b) methods of locomotion, (c) variation in shapes.

Second Day: *Measuring and classifying plankton:* (a) obtaining representative measurements of dominant forms (b) identifying common types.

Third Day: *Total Sample Analysis:* (a) learning how to count plankton (b) estimating size of population in comparison to total volume of water sampled.

TO THE STUDENT

There exists in the oceans of the world, in seas, bays, lakes, and in nearly every other natural water body, a population of organisms so immense that it defies counting! Everyone, while swimming has probably brushed up against millions of these creatures without being aware of their presence. No doubt some even swallowed a large number while learning to keep mouths closed underwater! Although limited studies of this population were made before 1887, it was not until that year when the oceanographer, Victor Hensen, first proposed a name for this vast assemblage . . . *plankton.* The term refers to those plants and animals mostly microscopic in size, that are made "*to wander or drift*" (Hardy, 1961), under the influence of ocean currents and tides. Even though many planktonic forms have the ability to swim, their efforts in the presence of oceanic water movements are generally too feeble and in vain. Animal members are named *zoo plankton*.

With the exception of marine mammals and reptiles, nearly every creature in the sea spends either a part or all of its life drifting about. Eggs, larvae, and juveniles of most invertebrates and fishes and even some adult forms are common. *Copepods* (crustaceans) are the most abundant and universally distributed animals in the plankton.

Phytoplankton (plants) are more numerous than their animal counterparts, and are best represented by the microscopic *diatoms* that form the vast bulk of the ocean's vegetation.

Because of their numbers, wide distribution and beneficial biological activities plankton are considered the most important inhabitants of the marine world with all forms of life directly or indirectly dependent upon them. Plankton are basic to the food chains of all marine life. Sponges, tube worms, clams and sea squirts filter out sea water to gather them. Herring fisheries use plankton indicators to predict the potential catch (Russell and Yonge, 1963). Giant baleen whales, over 100 feet in length and reaching fantastic weights of 150 tons, feed exclusively on plankton (Pequegnat, 1958). Diatoms are an important source of vital oxygen and proteins which animal life cannot synthesize but require. Without plankton the seas would surely be a wet desert!

OVERALL PURPOSE

The purpose is to observe and analyze a plankton sample taken from ________________________________.

location

MATERIALS

compound microscope
dissecting scope
petri dish
preservative (3-5% formalin)
centimeter grid or graph paper
sample bottle and label
plastic metric ruler
eye dropper
standard slides and cover slips
concavity slides
diluted plankton sample for each team

GENERAL INSTRUCTIONS

1. This laboratory study will extend for three periods.
2. Procedures are divided into daily activities.
3. Work as a team.

First Day ________________________________

Date

PURPOSE

To observe living plankton.

PROCEDURE

1. The teacher will provide the recorded conditions under which the plankton sample was made. Enter this information on the data sheet provided.

2. Obtain diluted portion of the plankton sample and pour into a petri dish. Gently swirl the contents to distribute.

3. Observe with a dissecting microscope. Scan the entire field.

4. Many organisms are too small to be seen with the dissecting scope. Prepare wet mounts of the sample and observe under a compound microscope with low and high power objectives. Do not discard any part of the sample. Empty wet mounts back into petri dish. Rinse with medicine dropper of water.

5. *Do not record any* observations today. Look for:

   a. most abundant organisms
   b. variations in shape, color and swimming abilities
   c. types of appendages
   d. chlorophyll-containing organisms
   e. eggs
   f. larval and juvenile forms of crustaceans and fish (see pictorial guide to the plankton, pages 68-71).

6. Preserve sample in 3-5% formalin before leaving. Label sample bottle.

Second Day ________________
Date

PURPOSE

To draw, measure and record characteristics of dominant organisms in plankton sample.

PROCEDURE

1. Select the most common organism from your preserved sample. Prepare a wet mount and view with low power (or high power).

2. Record the following information on data sheet:

   a. a detailed penciled drawing of the specimen;
   *b. measured actual size, in microns
   *c. measured drawing size, in microns
   d. magnification of drawing
   e. identification

3. Repeat this procedure with as many different specimens as time permits. *Record all observations.*

4. Do not discard any portion of your sample. Empty wet mounts back into sample bottle. Rinse with dropper of water.

Third Day ________________
Date

PURPOSE

To complete drawings, measurements and observations on preserved plankton and to compute the size of the plankton population in comparison to total volume of water strained.

1. Complete additional drawings, measurements and observations as required.
2. Compute total volume of water strained.

BACKGROUND

A plankton net with a measured net opening was towed behind a boat for a measured distance. How much water passed through this net? The total number of organisms collected from this tow represents the concentration of plankton in *that* volume of water.

a. To compute water volume which passed through net *towed behind a boat*:

$$M^3 = \frac{\pi D^2}{4} \times L$$

M^3 = volume of water in cubic meters
D = diameter of net in meters
L = length of tow in meters

for example: a net with a diameter of 2 decimeters was towed 1500 meters.

$$= \frac{3.14 \times .2^2}{4} \times 1500$$

= 47.1 cubic meters

b. To compute volume of water which passed through net *tossed from a bridge*:

** See Lab Exercise, Measuring with a Microscope – page 20.*

first determine length of tow:

$$L = \frac{Ti}{t} \times W$$

L = length of tow in meters

Ti = length of time net was immersed in water

t = time floating object took to pass width of bridge

W = width of bridge in meters

for example: a net strained water for 18 minutes. It took a cork .25 minutes (15 seconds) to pass under a bridge 12 meters wide. Thus

$$L = \frac{18}{.25} \times 12$$

L = 864 meters

... *now compute water volume* (assuming that net diameter is still .2 meters).

$$M^3 = \frac{\pi D^2}{4} \times L$$

$$= \frac{3.14 \cdot .2^2}{4} \times 864$$

= 27.1 cubic meters

3. Compute the density of the original concentrated sample (number of macroscopic specimens/__________ml).

BACKGROUND

The original sample contained _______________ml of liquid. It was diluted with an equal volume of sea water. You then received ____________________ml of this diluted sample. How many macroscopic (visible) organisms are contained in: (a) your sample and; (b) the original sample. Diatoms and other microscopic forms have been eliminated from this count.

a. Pour your sample into a petri dish.

b. Determine the area of the dish.

$$A\,(cm^2) = \frac{\pi D^2}{4}$$

A = number of sq. centimeters covered by dish.

c. Place petri dish over a centimeter grid or graph paper with centimeter squares clearly marked. Distribute sample evenly over bottom of dish.

d. Select, *at random*, five (5) squares and count all macroscopic organisms in each. To determine *average* divide total number of organisms by 5.

e. To estimate total number of organisms in the petri dish, (thus in your sample):

$$T = \frac{t}{g} \times A$$

T = total macroscopic organisms in your sample

t = total count from random grids

g = number of grids counted

A = area, in cm^2 of petri dish

for example: 100 organisms were counted from 5 random grids. The area of the petri dish covered 78.5 cm^{2}2.

$$\text{thus} = \frac{100}{5} \times 78.5$$

= 1570 macroscopic organisms/ ml diluted sample.

f. Compute total of macroscopic organisms in original concentrated sample:

$$Q = T \times \frac{V}{v}$$

where...

Q = total macroscopic organism passing through plankton net.

T = total number of organisms in your sample.

V= total volume of original concentrated sample.

v = volume of your sample

for example: Ten (10 ml) of sample was strained from 30 cubic meters (M^3) of sea water. You received 1 ml of this and proceeded to count 1570 macroscopic organisms. Thus:

$$\frac{1570 \times 10}{1} = 15{,}700 \text{ macroscopic organisms/ } 30\ M^3$$

g. Compute the total number of macroscopic organisms per cubic meter.

4. Record all information on data sheet.

PLANKTON LABORATORY

Data Sheet

I. Record the following conditions pertaining to the sample under study:

a. date____________________

b. location__________________

c. time of day________________

d. tide____________________

e. depth___________________

f. tow time_________________

g. mesh size________________

h. diam. net opening__________meters

i. distance net towed__________meters

j. dilution of your sample_______

k. volume of your sample_______ml

II.

organism #	1	2	3	4	5	6
drawing						
actual size						
drawing size						
magnification						
identification outstanding features+? coloration? common and/or scientific name						

III. a. volume of water which passed through the plankton net __________________________________M3

b. number of macroscopic organisms in your sample (T) __________________________________

c. number of macroscopic organisms in original sample (Q) __________________________________M3

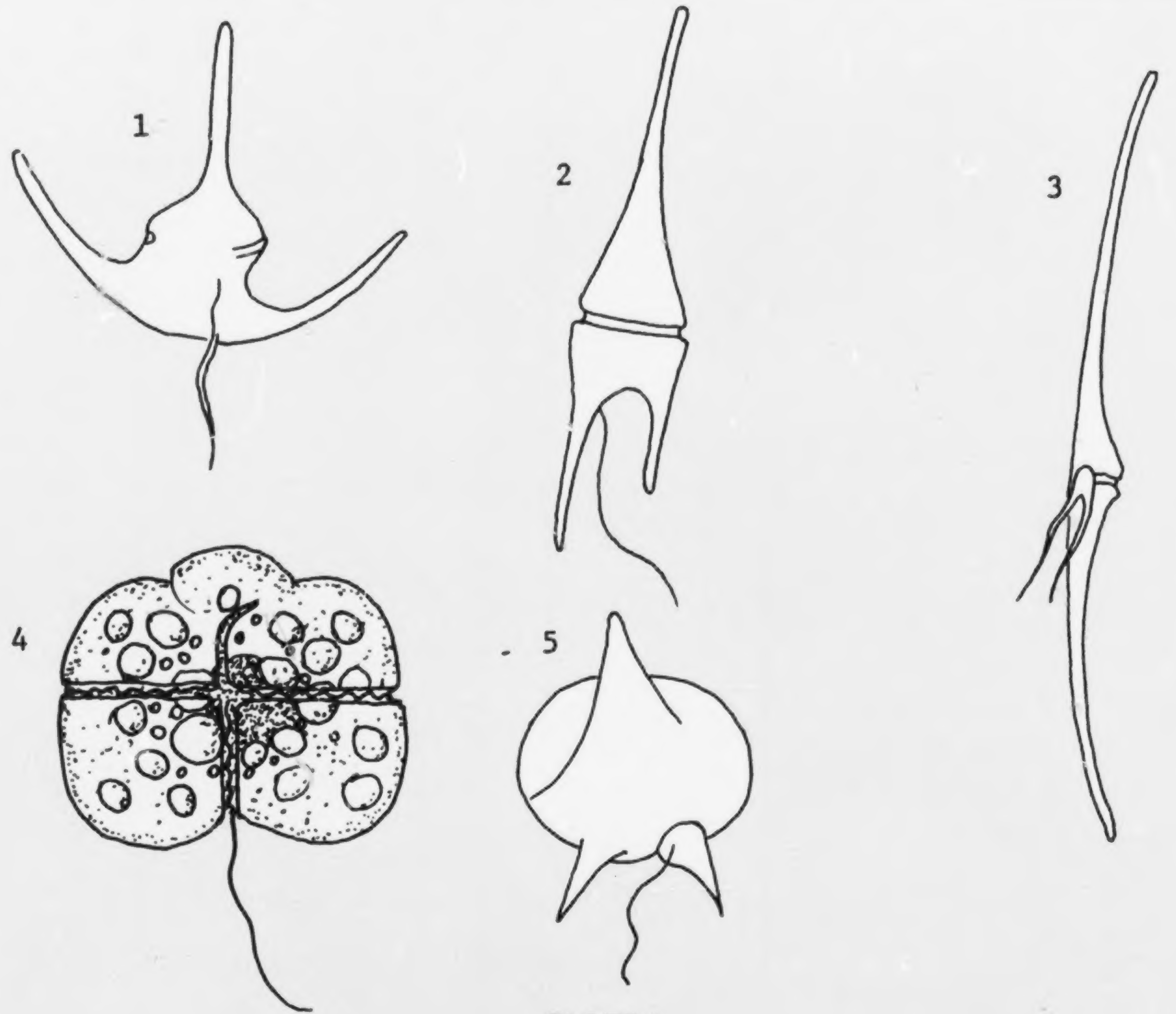


PLATE I

Dinoflagellates:

1. **Ceratium tripos**, x 200
2. **Ceratium furca**, x 400
3. **Ceratium fusus**, x 200
4. **Gymnodinium brevis**, x 1700
5. **Peridinium depressum**, x 200

1, 3, 4, 5 from Russell & Yonge (1963); 2 from Hardy*

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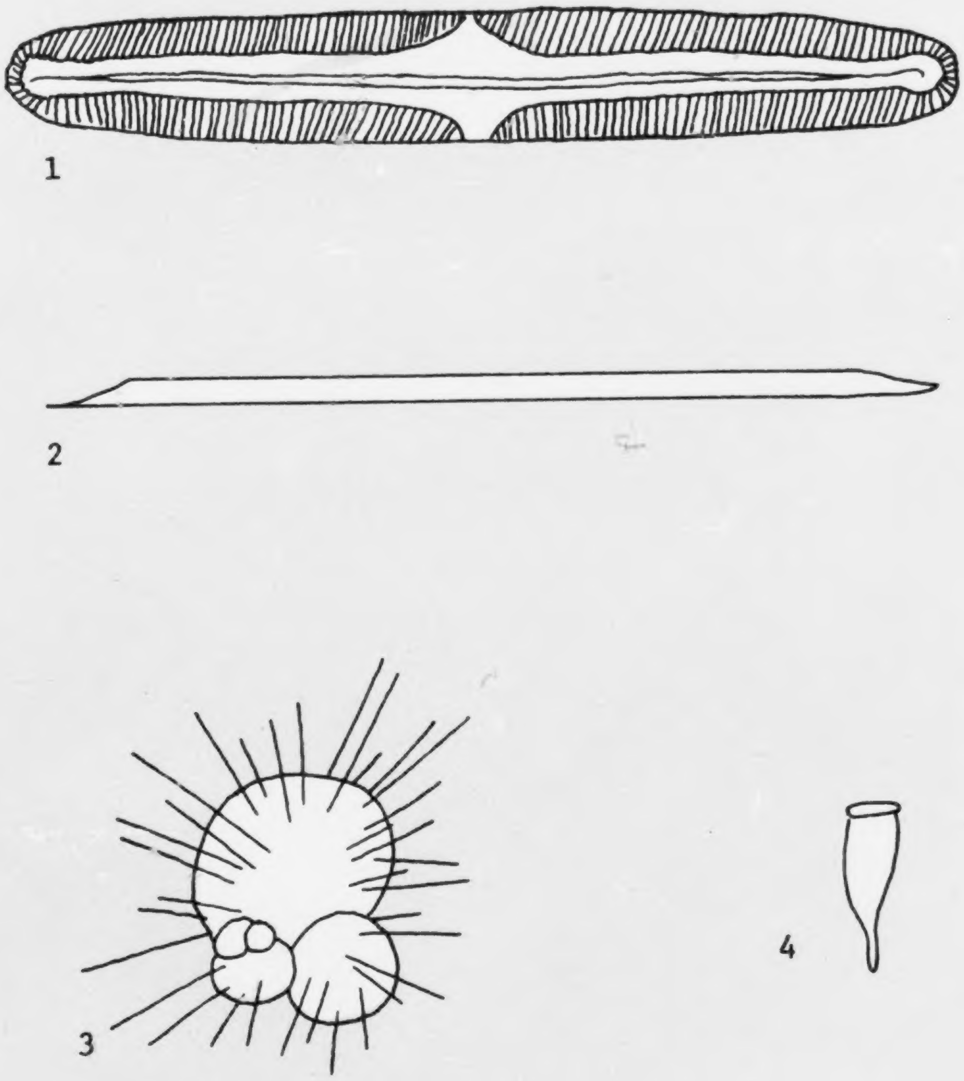

PLATE II

1. Naviculoid diatom, **Pinnularia sp.**, x 1200 (Chrysophyta)
2. **Rhizosolenia sp.**, a diatom, x 400 (Chrysophyta)
3. **Globigerina sp.**, a forameniferan, x 120 (Protozoa)
4. Tintinnoidea, x 120 (Protozoa)

1, 3, 4 from Fraser (1962) 2 from Hardy.*

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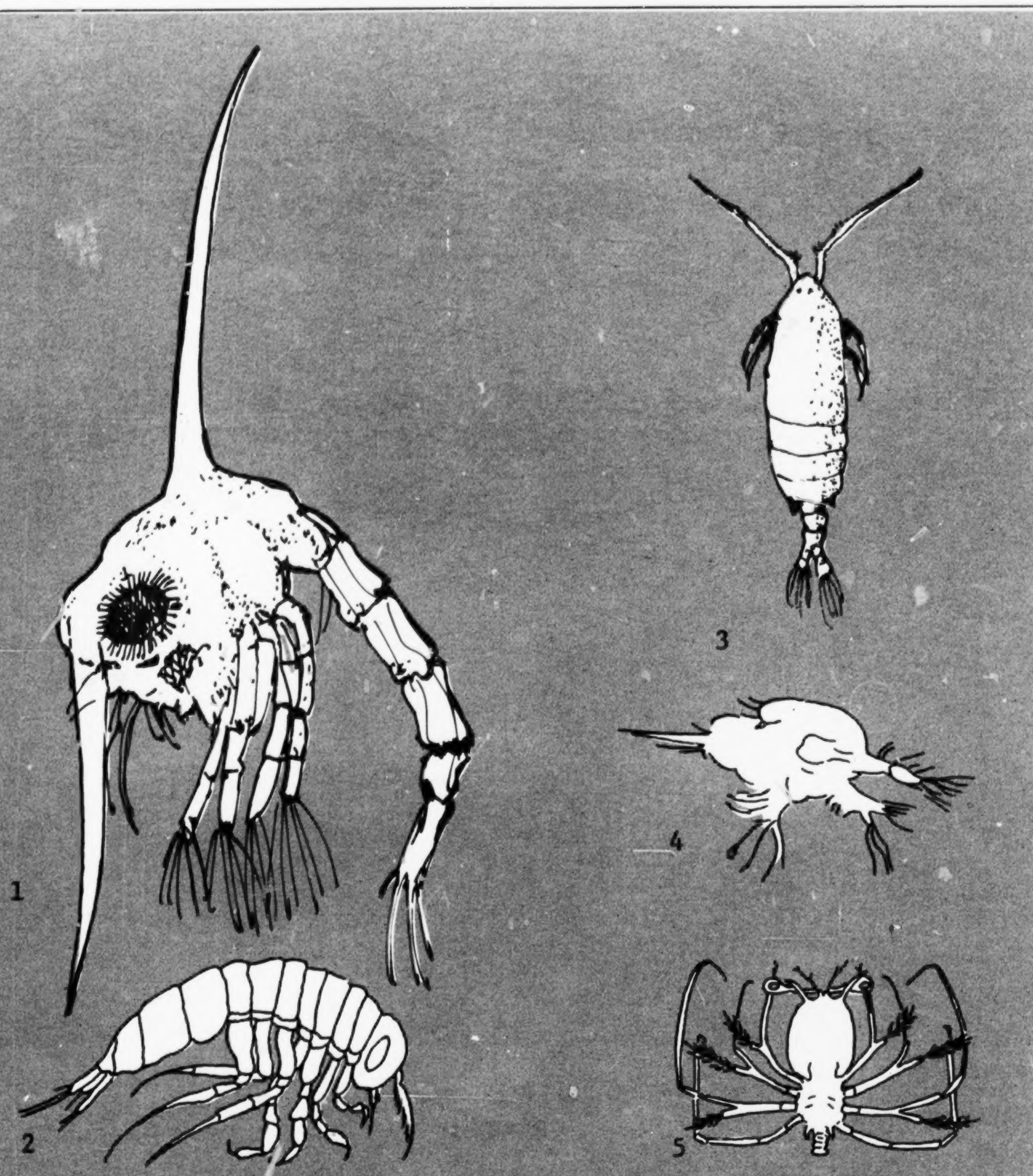

PLATE III

1. Zoea larva of crab, x 40 (Arthropoda)
2. Amphipod, x 5 (Arthropoda)
3. Copepod, x 15 (Arthropoda)
4. Nauplius form of copepod, x 80 (Arthropoda)
5. Phyllosoma of spiny lobster, x 1-1/2 (Arthropoda)

All above from Fraser (1962)*

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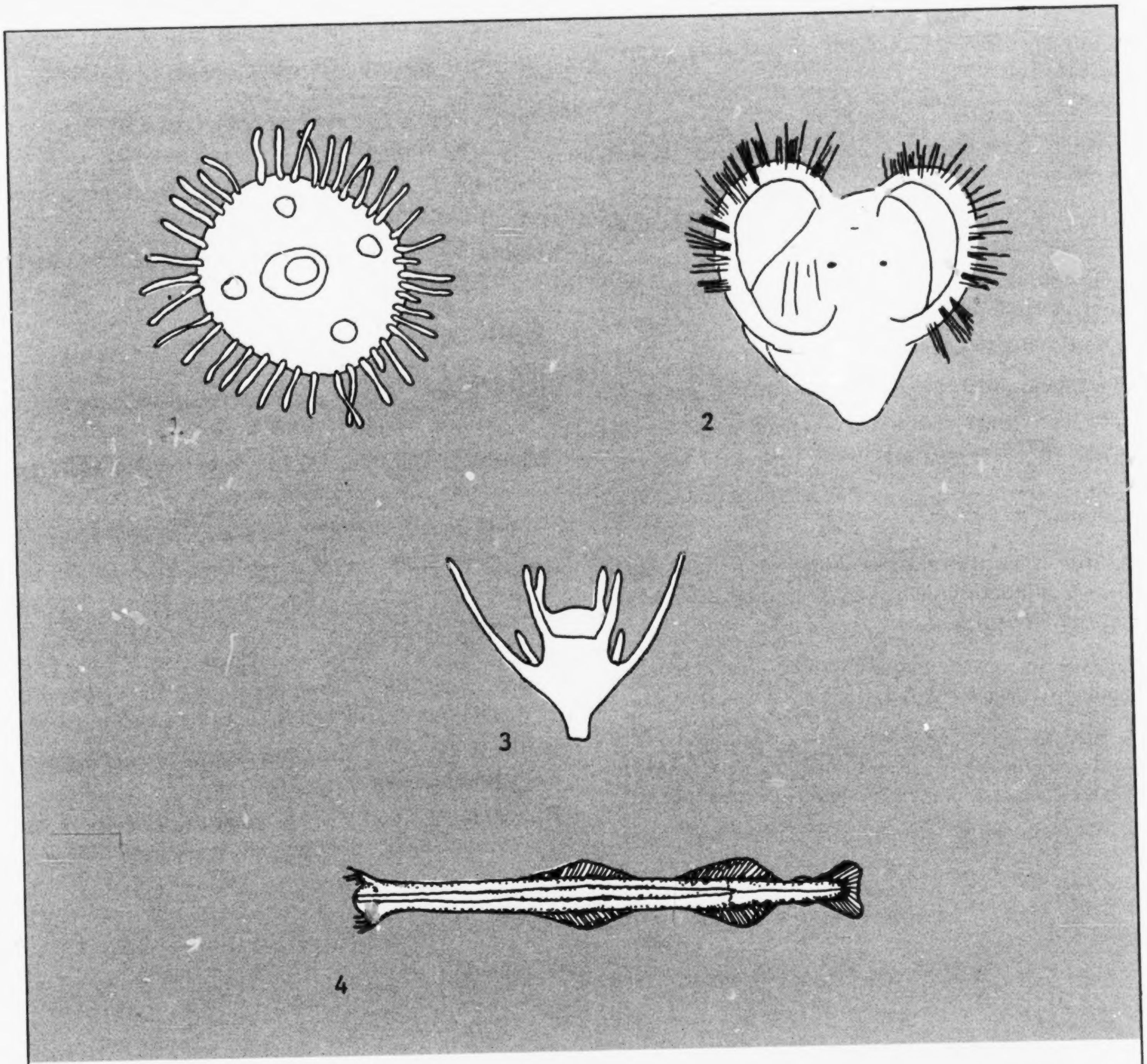

PLATE IV

1. Medusa of the hydrozoan, **Obelia sp.**, x 17 (Cnidaria)
2. Veliger larva of a gastropod, x 60 (Mollusca)
3. Pluteus larva of a brittle star, x 50 (Echinodermata)
4. Arrow worm, **Sagitta sp.**, x 5 (Chaetognatha)

1, 2 from Hardy, 3, 4 from Fraser (1962)*

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QUESTIONS FOR CONSIDERATION

1. How are organisms which are not part of the plankton population classified?

2. What is the DSL?

3. Which environmental factors may influence the vertical migrations of plankton?

4. How do minute and delicate plankton withstand the crushing pressures of deeper waters?

5. How do diatoms and copepods figure into the food chains of marine organisms?

6. How do baleen whales feed on plankton?

7. Are there ocean areas in the world devoid of plankton?

8. What was your impression of the abundance of diatoms in the plankton sample? Copepods?

9. Which phyla of organisms were best represented in your sample?

10. What happens to plankton which "drift" into waters where conditions of salinity, oxygen, or temperature are unfavorable? Explain answer.

11. What are some limitations to using a plankton net in sampling a population of organisms?

12. What are some limitations to the methods used in counting the density of plankton populations? What are other methods of estimating the density of plankton?

13. What is meant by "standing crop of plankton"?

14. What do the prefixes, "holo-, mero-, and nano-" refer to in reference to plankton? Give examples.

15. What adaptations do plankton have for moving?

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THE LIVING WORLD WITHIN A SPONGE

TO THE TEACHER

This study will help students realize one important role of sponges in the marine environment . . . that of providing shelter for a horde of creatures.

Fresh Loggerhead sponges are recommended for this activity. Speciments may be collected one day, enclosed in strong plastic bags, stored in a refrigerator overnight and used the next day. Further delay will prove highly undesirable . . . sponges decompose rapidly!

Small sponges could be assigned to one student, while larger specimens would prove enough material for a team or even an entire class.

TO THE STUDENT

Sponges are strange creatures and not at all like the typical animals that are familiar. They have no head, body, arms, legs or any other obvious structures that are generally associated with animals. All that is apparent are the countless pores which riddle their strong-smelling tissues.

Sponges are microscopic during their larval stage and drift about as part of the plankton population. Their wanderings are short-lived, as they soon settle to the bottom and become anchored . . . destined to spend the rest of their days in one place!

Although no animal looks to the lowly sponge as a source of food (try tasting a sponge sometime), many creatures take up residence in their intricate network of canals. Rachel Carson (1955) wrote:

> "One such permanent lodger is a small shrimp – one of the group known as snapping shrimp because of the sound made by snapping the large claw. Although the adults are imprisoned, the young shrimp, hatched from eggs adhering to the appendages of their mothers, pass out with the water currents into the sea and live for a time in the currents and tides, drifting, swimming, perhaps carried far afield. By mischance they may occasionally find their way into deep water where no sponges grow. But many of the young shrimp will in time find and approach the bulk of some loggerhead sponge and, entering it, will take up the strange life of their parents. Wandering through its dark halls, they scrape food from the walls of the sponge. As they creep along these cylindrical passageways, they carry their antennae and their large claws extended before them, as though to sense the approach of a larger and possibly dangerous creature, for the sponge has many lodgers of many species – other shrimps, amphipods, worms, isopods – and their numbers may reach into the thousands if the sponge is large."

PURPOSE

To discover which types of organisms are residents of sponges.

MATERIALS LIST

dissecting kits
sorting jars
dissecting microscopes
balances and rulers

PROCEDURES

1. Carefully slice off a thin section of sponge tissue. Examine for animal life.
2. Sort out creatures according to phyla, class, etc.
3. Repeat this procedure until entire sponge has been dissected and all inhabitants removed and sorted.

SUGGESTED MATHEMATICAL COMPUTATIONS

1. Record total population found in sponge.
2. Compute per cent of total population for each species by numbers and weights.
3. Compute weight-length ratio for each taxonomic group.

SUGGESTED GRAPHIC REPRESENTATION OF DATA

Prepare a graph showing the following:

1. Frequency distribution of sponge population.
2. Weight distribution according to species.

QUESTIONS FOR CONSIDERATION

1. How could one account for the presence of animal life within a living sponge? How would you classify this relationship?

2. What kinds of competition would go on between the tenants?

3. Is the sponge harmed in any way because of these tenants?

4. Can you attach any economic importance to the sponge's role of providing shelter for a multitude of creatures?

5. Do you think that the population inhabiting a sponge changes with seasonal conditions? Why?

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SPONGE SPICULATION

INTRODUCTION

The body of a sponge is a loose aggregation of cells held together with spicules or spongin (or both) which form a framework. Sponge classification is often based on the shape and chemical constitution of the spicules.

TO THE TEACHER

A. To distinguish between calcareous and siliceous spicules: This lab deals primarily with spicule forms. After the spicules have been isolated onto the glass slide, separate the calcareous types from the siliceous types by adding a drop of 1N HCl. The calcareous spicules will dissolve, the siliceous spicules will remain.

B. An alternate lab method: (one class period – 50 minutes) Some sponge tissue can be dissolved in sodium hypochlorite (Clorox). Use a depression slide, then transfer to a regular slide for further examination. Some residue of sponge tissue may remain with this method, but spicules can be observed, and the technique involves less time.

C. The lab using KOH will take some students more than one lab period. Some will save the filter paper and do observations on the second day.

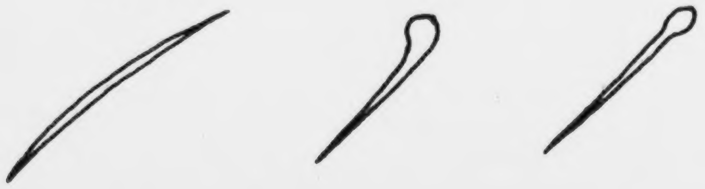
D. Safety goggles are indicated when KOH is used.

TO THE STUDENT

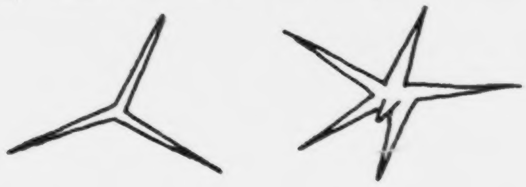
Spicule forms are:

Spicule forms are:

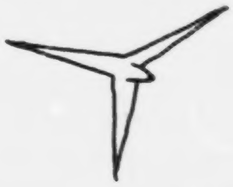
(A) Monaxon – single axis, like a stick, sometimes pointed; one end curved or club-shaped, depending on species.



(B) Triaxon – in three of multiples of three



(C) Tetraxon – four-pronged



(D) Polyaxon – many-pronged

Sponges are also classified by the material making up spicules:

(A) Calcareous (contains calcium; $CaCO_3$)

(B) Siliceous (contains silicon; SiO_2)

(C) Sponges with both kinds

This phylum is represented by three classes:

(A) Class Calcarea – spicules are exclusively calcareous; monoaxon or tetraxon

(B) Class Hexactinellida (the glass sponge) – spicules are siliceous; tetraxon

(C) Class Demospongiae – contain spongin; spicules are siliceous

PROBLEM UNDER INVESTIGATION

To study the characteristic spiculation of sponges.

SPECIAL INSTRUCTIONS FOR THE STUDENT

In order to dissolve the body substance of the sponge samples it is necessary to use a base such as potassium hydroxide or hydrogen peroxide. Bases are quite caustic. Exercise care.

MATERIALS

Alcohol lamp, matches or a lighter

Ring stand with ring
Funnel and two pieces of filter paper
Test tube rack
Two test tubes, each tube with 2ml of KOH 20%
Test tube holder
Compound microscope
Two glass slides and cover slips
Bumping beads
Dissection kit
Beaker or baby-food jar to catch filtrate
Wash-bottle with distilled water

PROCEDURE

A. There are two different kinds of sponges to be examined. From each sponge, cut off a piece about 5mm x 5mm. Cut each of these into *very* tiny bits. Place these little particles *separately* into the test tubes containing the KOH. Add two glass bumping beads to each tube.

B. Wear safety goggles!

C. Using the test tube holder, heat the test tube and its contents very slowly over the alcohol burner. Repeat – gently; slowly!! Don't overboil! Keep the test tube pointed *away* from people! Continue heating gently for 8 minutes.

D. At the end of 8 minutes, allow the tube to cool for another minute, then fill it nearly full with distilled water.

E. Having already set up the filtering apparatus, pour the entire contents of the tube through the filter paper. As soon as most of the liquid has run through the filter paper, follow this up with another tube – full of distilled water. This is to rinse away the KOH. Save the bumping beads.

F. When all the liquid has run through, place a tiny bit of the residue left on the filter paper onto a glass slide. Add a drop of distilled water and a cover slip and examine under the microscope.

G. It may be better to cut down the light on the microscope since the spicules themselves are practically transparent.

INSTRUCTIONS FOR RECORDING DATA:

A. Describe the original piece of sponge

B. Draw the spicules seen

C. *Estimate the length of spicules in microns:

1. Sample #1 ____________________ microns
2. Sample #2– ____________________ microns

Sample #1 Sample #2

D. Suggested Graphic Representation of Data.**

QUESTIONS FOR CONSIDERATION

1. What is meant by "characteristic" spiculation?
2. What type of habitat would be best suited to an animal with this type of body structure?
3. Would this kind of body structure be suitable for a free-swimming animal?
4. Would there by any limitations as to the size of an animal with this kind of body structure?

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**See "Measuring With a Microscope" – page 19*

***See "Statistical Analysis"*

STINGING CELLS - PHYLUM CNIDARIA [Coelenterata]

INTRODUCTION

Especially abundant in the tentacles are specialized cells called cnidoblasts which contain stinging structures called nematocysts. Possession of nematocysts is a characteristic peculiar to the phylum. According to Barnes (1963): "Nematocysts, which can be discharged from the cnidoblast are used for anchorage, for defense, and for the capture of prey."

TO THE TEACHER

Use of this lab depends somewhat on the availability of live specimens. One or two specimens of adult (medusa) Scyphozoans (jelly-fishes) such as *Aurelia sp.* will suffice for 75 students. Time required: one lab period – 50 minutes. These animals, kept alive in well-aerated aquaria, will remain alive for three or four days despite frequent tentacle, cutting. *Physalia,* the Portuguese man-of-war, should be avoided for reasons of safety especially so far as the student collector is concerned.

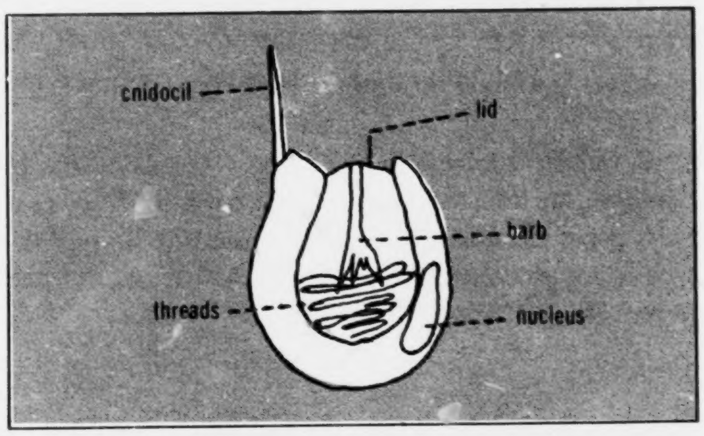
Small fish have been considered as symbionts among the tentacle structures. *Nomea* and other animals have special adaptations for eating the stinging cells of the host. These must be considered parasites.

TO THE STUDENT

Nematocysts in the Cnidaria (nid-ar'-e-a) appear in many different forms, as well as exhibiting different actions. According to Hyman (1940), R. Weill has recognized seventeen different types of nematocysts, based on the characters of the discharged tube. Dr. Hyman also states: "Nematocysts can be used only once and after discharge are cast off."

The type of nematocyst used to wrap around and entangle is called a volvent; those that are usually barbed are called penetrants; and the type containing a toxin which is released through an open end on the ejected tip are called glutinants.

The cnidoblasts you will see are very small. "Most capsules range between 5 and 50 $m\mu$ ($m\mu$ = 1 millionth mm.) in length and discharge a tube so fine that the details of spination are often very difficult to ascertain." Hyman (1940) A "typical" cell (nematocyst within a cnidoblast) will look like this, undischarged:



Look for discharged nematocysts by finding the long thread leading away from an empty cell.

It is possible to witness the discharge of a nematocyst. Be on the watch for this. Nematocysts are somewhat "tem-permental" when one tries to induce a discharge. "The discharge or explosion is believed to result from the proper stimulation of the cnidocil by food, prey, or enemy animals. Both chemical and mechanical factors appear to be involved." Hyman (1940).

PROBLEM UNDER INVESTIGATION

The problem in this exercise is to view stinging cells (nematocysts) from viable jellyfish tentacle tissue under the microscope. Observations are to be made upon cells which have not discharged, those that have discharged, and perhaps those in the act of discharging. The problem also involves collecting and maintaining live jellyfish specimens as well as techniques in wet-slide preparation of living tissue.

MATERIALS

compound microscope
microscope slide and coverslip

dissecting kit
dilute Methylene Blue stain
one-half piece of filter paper

PROCEDURE

A. From the living scyphozoan, snip off a small (0.5cm) piece of tentacle with scissors and forceps. Be gentle *and* be quick!! Place the specimen on a clean glass slide. Mascerate (cut up) this tissue with the tip of the scalpel blade.

B. After adding a drop of distilled water and a cover slip, observe under low power. Try to find both the "exploded" and "unexploded" cells; observe also under high power.

C. For added emphasis, add a drop of Methylene Blue by "pulling" the stain through under the cover slip. A piece of filter paper placed at the edge opposite the drop of dye will accomplish this. Observe again under low and high powers. Be on the watch for "exploding" cells!

INSTRUCTIONS FOR DATA RECORDING:

A. Sketch the following:
  1. An "unexploded" cnidoblast
  2. An "exploded" cnidoblast

B. Determine the following: (See microscopic measurement on Page 19).
  1. Width of a cnidoblast, in microns
  2. Length of an "exploded" thread, in microns

QUESTIONS FOR CONSIDERATION

A. What distinguishing physical characteristic separates the Phylum Cnidaria from the Phylum Ctenophora? Historically, Phylum Coelenterata used to include the sponges, coelenterates and ctenophores!

B. What other cnidarians bear watching when one is skin diving?

C. What form of cnidarian would be associated with terms such as "fringe", "barrier", and "atoll"?

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THE PELECYPOD GILL
(Phylum Mollusca)

TO THE TEACHER

The quahog, *Mercenaria sp.*, and other Venus clams (Family Veneridae), are relatively easy to obtain, and represent a high level of pelecypod evolution. From the standpoint of bay pollution, the position of the clam is unique. The ciliary feeding habit causes this animal to accumulate quantities of bacteria, pesticides, and radioactive materials beyond the standards set for human consumption. Thus, we see a direct economic influence of pollution upon a source of food from the sea.

Given adequate aeration, five or six quahogs are sufficient for this exercise. They can be maintained a week or so in the lab.

You may choose to open the valves yourself, but if time and quantity of specimens allow, it is interesting for the students to open their own clams. Care should be exercised in the handling of sharp instruments.

TO THE STUDENT

The gills of the pelecypod serve a dual purpose: (1) to perform the function of oxygenation and (2) to move food particles toward the food groove. The term "ciliary-mucoid feeder" is applicable to the pelecypod, as well as to other marine groups, since the action of cilia combine with adhesive mucus secretions in the nutrition processes. While the combination of cilia and mucous is present in other portions of the digestive tract, the action of cilia can be observed, without difficulty, in gill examination.

A. In collecting clam specimens, keep in mind the type of biome where these animals are found. Even though pelecypods inhabit other biomes, they are most certain to be found along with other burrowing animals, especially with the segmented worms: hence, the term "Pelecypod-Annelid" biome which alludes to mud, estuaries, little wave action, exposure to the sun (insolation) for long periods, and lower salinity from fresh-water streams.

B. Notice how the ciliary-mucoid-filter-feeding method is carried out by a pelecypod. In opening the clam, be cautious in the use of any sharp instruments.

C. Circulation of water (and the particles therein) occurs as valve action moves the water in through the incurrent siphon, over and under the gills and exits through the excurrent siphon. In this exercise, the gill tissue, with its cilia, can be seen.

PROBLEM UNDER INVESTIGATION

To observe the location and arrangement of a pelecypod gill and to study its ciliary action by microscopic examination.

MATERIALS

Clam or quahog specimen (live)
Dissecting kit
Dull knife, spoon, oyster knife (optional)
Microscope slide and cover slip
Compound microscope
Methylene Blue stain (optional)
1/2 piece of filter paper (optional)

PROCEDURE

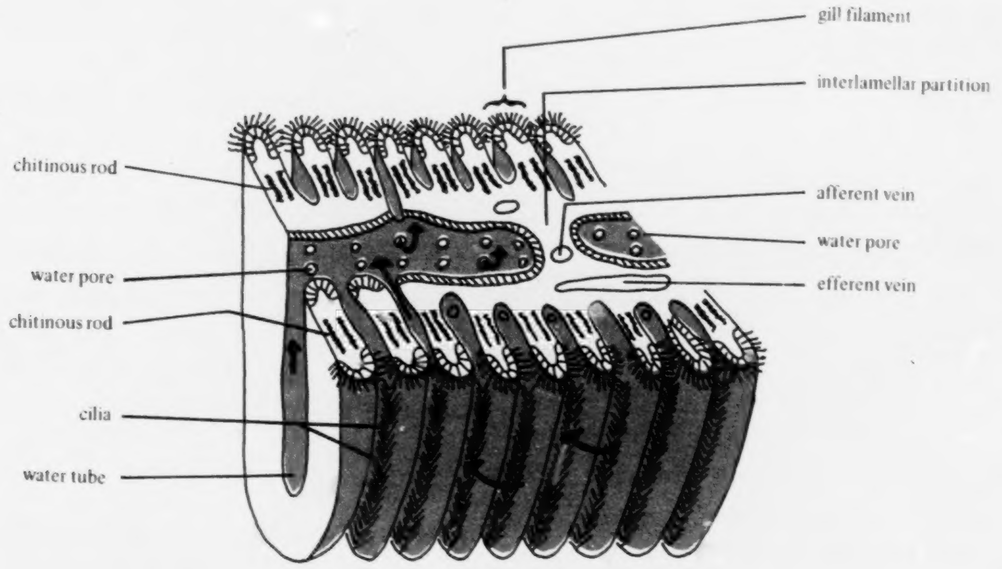
A. Dissect the valves by cutting the tendons holding the hinge and by cutting the muscles holding the valves. Proceed with caution!

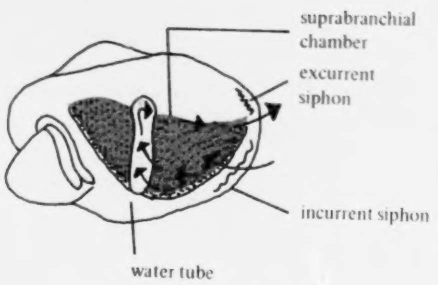
B. Note the arrangement of the gills, location and action of the heart and other organs.

C. Cut a piece of gill tissue (4mm x 4mm), place it on a microscope slide, add a drop of clean salt water and a cover slip, and observe under low and high powers.

Respiratory system of a clam:

Horizontal section through a fill showing arrangement of gill filaments, blood vessels, and water tubes. Arrow shows direction of water currents.


gill filament
interlamellar partition
chitinous rod
afferent vein
water pore
efferent vein
water pore
chitinous rod
cilia
water tube


suprabranchial chamber
excurrent siphon
incurrent siphon
water tube

A Whole Gill

D. After observations of ciliary movement are made, a drop of Methylene Blue may be added by "pulling" a drop of the stain through and under the cover slip. A piece of filter paper placed at the opposite edge of the cover slip will accomplish this.

INSTRUCTIONS FOR DATA RECORDING

Describe what you see and sketch a small portion of the gill tissue containing cilia.

QUESTIONS FOR CONSIDERATION

1. In your opinion, is the dissected gill "dead"?
2. How would you propose to measure the effect of temperature on the gill action of this animal. Tell what you would do. If there is time, you may try out your procedure.
3. By means of your own labeled drawing, show the following sides of a clam: dorsal side, ventral side, left side, and right side.
4. What other animals are "ciliary-mucoid" feeders?
5. Would you expect to find the "ciliary-mucoid" method of nutrition in free-swimming types of animals?
6. Describe how gas exchange occurs at the gill sites.

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HORSESHOE CRAB

TO THE TEACHER

This is a laboratory exercise requiring 1 to 3 periods depending upon the following: 1 period if only preserved specimens are available; 2 to 3 periods if both living and preserved specimens are available.

The horseshoe crab is a hardy animal. It can be collected several weeks or months ahead of the time planned for its use. It will survive with a minimum of care if fed an occasional piece of shrimp.

The horseshoe crab is found in the very shallow water of bays and estuaries, sometimes by the thousands in the spring and early summer.

TO THE STUDENT

The horseshoe crab is referred to as a "living fossil" since it has remained virtually unchanged over the past 400 million years.

The horseshoe crab (king crab) feeds on worms, mollusks, bottom-dwelling algae, plus other material it scavenges from sandy or muddy beaches and bottoms.

Dr. Talbot H. Waterman of Yale University's Osborn Zoological Laboratory has learned of the horseshoe crab's apparent ability to use a small patch of blue sky as a navigation aid. The horseshoe crab may be utilizing a system far superior to any system now used by man!

At one time the horseshoe crab was considered a pest along the upper east coast of the United States and a bounty of one cent was placed on each animal collected. The animals were then dumped on the town trash pile.

In other areas, they have been collected in great quantities and ground for fertilizer.

PURPOSE

To gain a knowledge of the external anatomy and behavior of an interesting Florida arthropod, *Limulus polyphemus*, the horseshoe crab.

MATERIAL

Preserved specimens
- dissecting pan or tray
- probe
- horseshoe crab

Living specimens
- aquarium
- light source
- small pieces of shrimp

PROCEDURE

Place the living horseshoe crab in an aquarium with a sand or gravel bottom for the following observations:

a. coloration of exoskeleton
b. method of locomotion
c. method of righting itself if turned upside down
d. attraction to light: The light should be oriented from a corner position to determine if there is a response
e. method of burrowing
f. number and kind of attached organisms
g. feeding habits
h. determination of sex
i. method of swimming

If preserved specimens are at hand, the student should locate and record a physical description of the external characters shown in figures 1 and 2 – pages 81, 82).

DATA RECORDING

Record all observations on the living specimen and repeat observations as many times as possible during one laboratory period, while checking for possible variations in behavior patterns.

FORMULA AND MATHEMATICAL COMPUTATIONS

Since the male genital openings are round and the female genital opening is a transverse slit, the number of males and females used by the class should be computed as percentages.

$$\frac{\text{Number of males}}{\text{Total number of specimens}} = \underline{\qquad\qquad} \%$$

GRAPHIC ANALYSIS AND CONCLUSIONS

It may be feasible in your area (providing horseshoe crabs are plentiful) to carry out one of the following suggested experiments:

1. survey the living population to determine how many specimens have a growth of algae or barnacles present and prepare a graph of size (carapace diameter) versus number of growths present.
2. map the locations on the body at which *Bdelloura* (a marine flatworm) occurs and compute the frequency with which it occurs at each point.
3. measure carapace diameter and telson length to be used in preparing a graph.

QUESTIONS FOR CONSIDERATION

1. Why and how does the horseshoe crab molt?
2. What function could the moveable spines located on the opisthosoma (abdomen) have?
3. What potential economic importance could this creature have?
4. What are the ancestral origins of the horseshoe crab?
5. What are the functions of the gnathobase of each appendage?
6. Do the median ridges offer protection for the compound eyes?
7. Does the complete body armor for internal organs and appendages account for the fact that they have remained unchanged over some 400 million years?
8. What predators must the horseshoe crab face?
9. What provides competition for the horseshoe crab in its environment?
10. Why do barnacles and algae seldom attach to the carapace of the horseshoe crab?
11. What navigation system does the horseshoe crab utilize?
12. Why does the horseshoe crab burrow in the sand?
13. Why is *Bdelloura* attached in the horseshoe crab?

LIMITATIONS

The range of the horseshoe crab is such that it is usually plentiful in South Florida and diminishes in numbers along the East Coast to Jacksonville. The West Coast of Florida has an adequate seasonal abundance.

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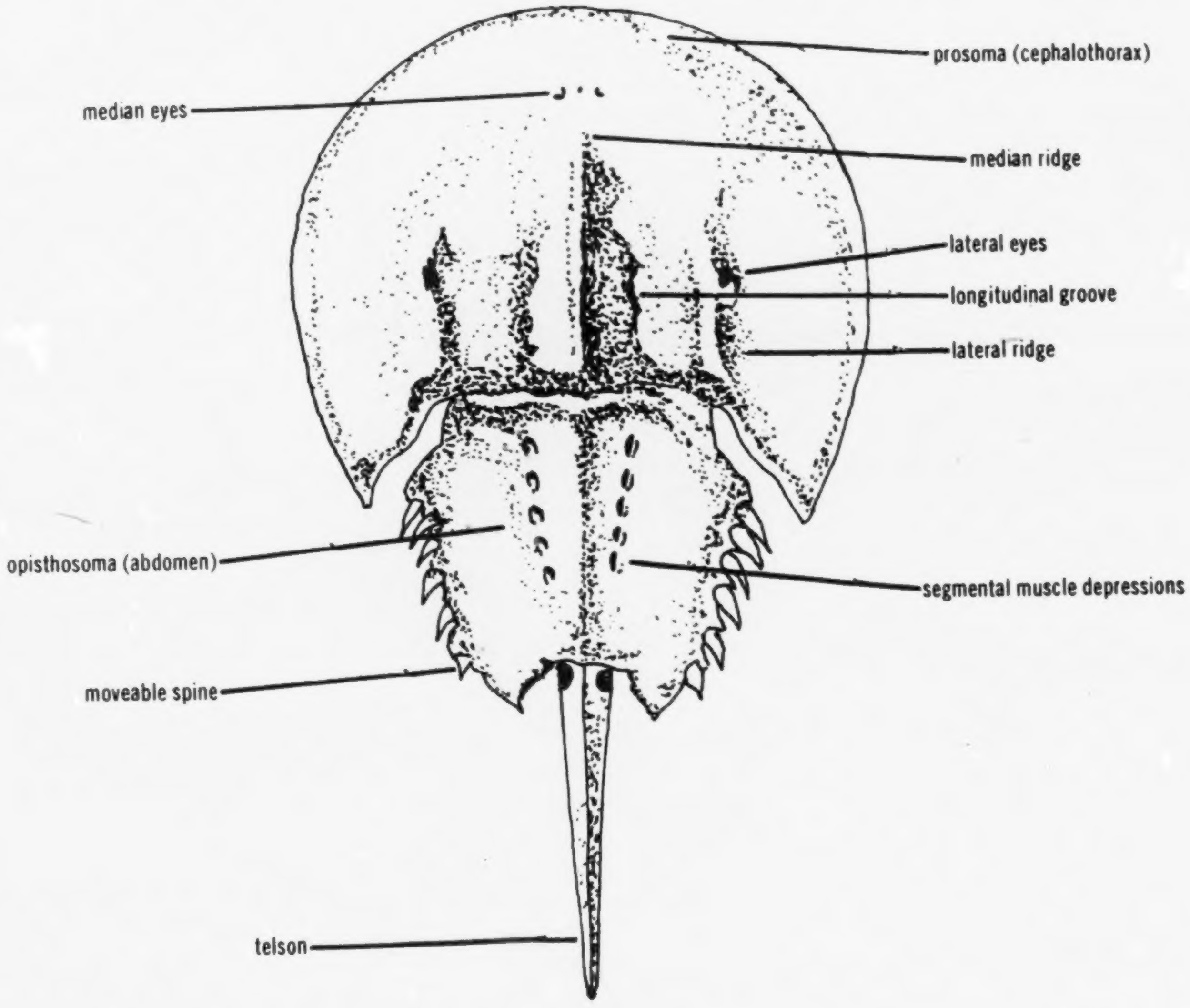
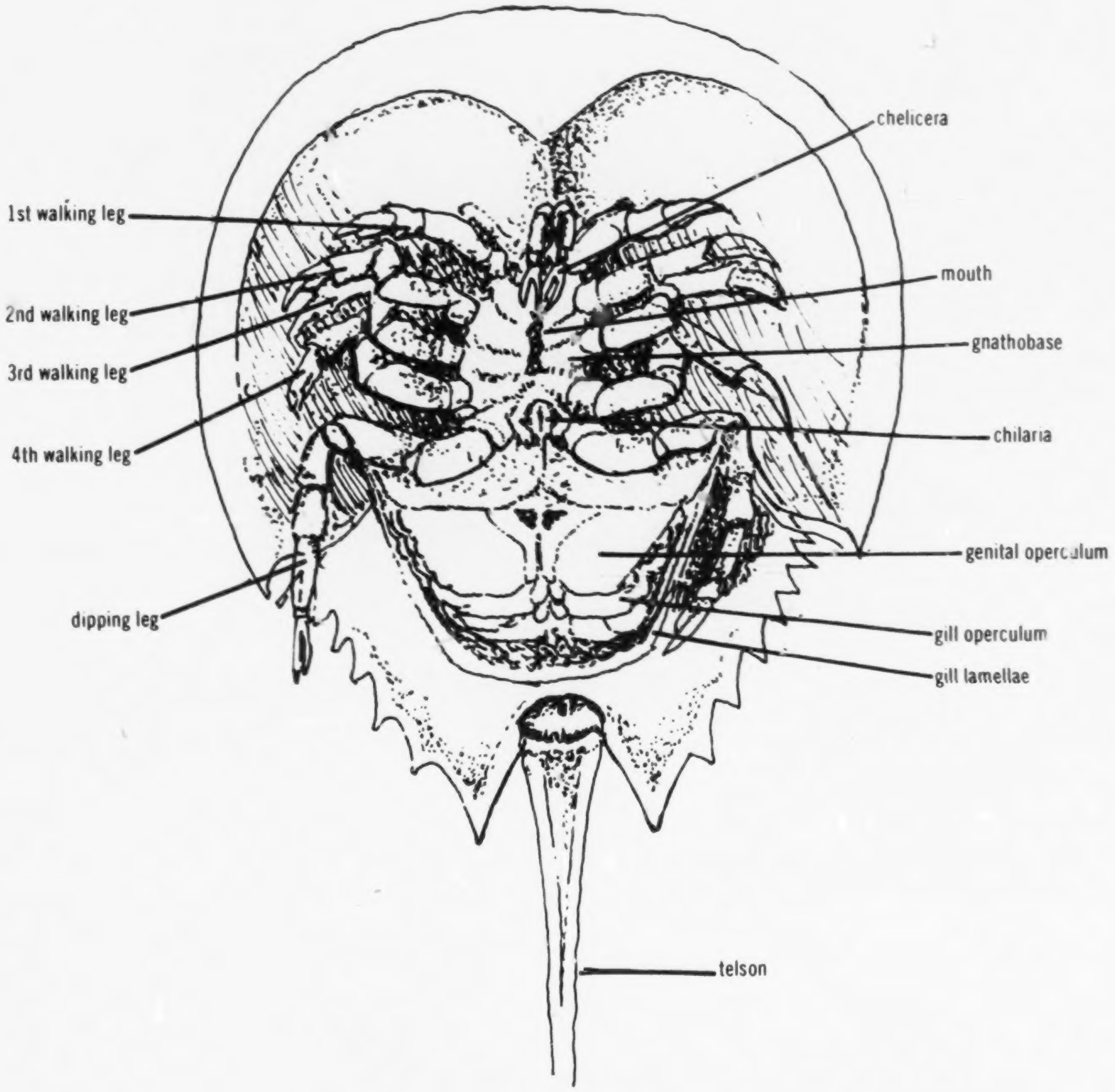

prosoma (cephalothorax)
median eyes
median ridge
lateral eyes
longitudinal groove
lateral ridge
opisthosoma (abdomen)
segmental muscle depressions
moveable spine
telson

DORSAL VIEW

FIG. 1


chelicera
1st walking leg
mouth
2nd walking leg
gnathobase
3rd walking leg
chilaria
4th walking leg
genital operculum
dipping leg
gill operculum
gill lamellae
telson

VENTRAL VIEW

FIG. 2

After Barnes Invertebrate Zoology figure 14-2, page 336.

INTRODUCTION TO THE MARINE ARTHROPODS: THE CLASS CRUSTACEA

The Phylum Arthropoda contains more species of animals than all other Phyla combined. Fortunately for the Marine Biologist only the Class Crustacea inhabits salt water. There are few exceptions to this – the pycnogonids, fantastically shaped "sea-spiders", some marine mites, the horseshoe crab, a few species of tardigrades (water-bears) and a few species of Collembolan insects. The relationship of these aberrant forms to other invertebrates is still obscure.

Since the literature concerning the Crustacea is so vast, only a few publications can be emphasized here. The first modern scientific monograph on the crayfish was by T. H. Huxley (1880). The impact of this book was so great that it created a trend in the teaching of invertebrate zoology. This method is the study of certain types. The difficulty with this method is that it does not give us an overall knowledge of any group. Those who do not make a career of biology all too often think that *paramecium* is representative of all protozoa, *hydra* is representative of all coelenterates, that the earthworn is the only Annelid and that that the Crustacea include only the crayfish, lobster and crab. The comprehensive textbook on the invertebrates by Barnes (1963) did much to give us a broad view of all invertebrates. Schmitt's *Crustaceans,* first published in 1931 and reprinted in 1965 should be in every science library. It is very readable and authoritative. The *Field Book of Seashore Life* by Roy Waldo Miner is invaluable in any marine taxonomic work.

In the classification of the Crustacea, the student has been given as complete a listing as possible, stressing the forms which are of the greatest economic importance. Crustacea are of quantitative importance in marine food chains. The sources of information are numerous (See references). Every Marine Science teacher is urged to obtain as many of the cited references as possible. A classroom reference library is indispensable. All Marine Science groups are urged to develop as large a study collection of preserved specimens as possible. The Crustacea of the beaches and off-shore waters of Florida are great assets – we need to know more about them. The aim is toward the beginning student of Crustacea taxonomy. The diversity of shape, structure, and function makes this lab both difficult and interesting.

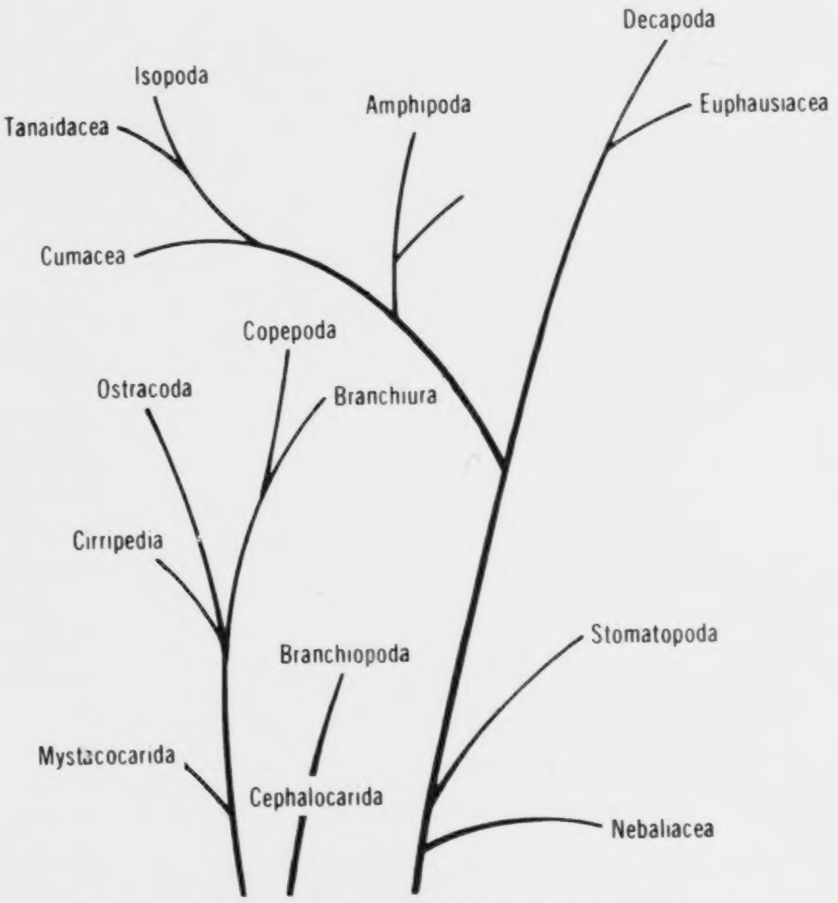
THE CLASS CRUSTACEA: SUMMARY OF MARINE SPECIES
(modified from Barnes and Schmitt)
26000 species

Subclass– No. of Species	Order– No. of Species	Characteristics and/or Habitat
1. *Cephalocarida* 3		Blind, colorless, minute, found in sand and mud of beach.

2. *Branchiopoda* Mostly fresh-water. Usually only a few millimeters in length. Pale and transparent.	Anostraca (175) Fairy Shrimp	Only a few marine species. Brine shrimp *(Artemia)* found in salt lakes throughout world.
	Notostraca (15) Tadpole Shrimp	
	Conchostraca (180) Clam Shrimp	Looks like a little clam.
	Cladocera (425) Water Fleas	A few marine forms, mostly fresh-water.
3. *Ostracoda* 2000 Mussel or seed shrimp, many marine forms.	Myodocopa (300)	Marine.
	Cladocopa (30)	Marine.
	Podocopa (1600)	Marine and fresh-water.
	Platycopa (30)	Marine.
4. *Mystacocarida* 4		Rare, minute, marine. Only a few ever found. Planktonic.
5. *Copepoda* 5000 Mostly marine but many fresh-water forms. Many parasitic. Exist in enormous numbers and most important link in food chains from whales to fish.	Calanoida (1200)	*Calanus*
	Harpacticoida (1200)	Benthic. Fresh & Salt. *Harpacticus* A few parasitic forms.
	Cyclopoida (1000)	Planktonic and Benthic. Fresh and salt. Cyclops. Many are parasitic.
	Notodelphoids (300)	*Poropygus*
	Monstrilloida (35)	Marine. Larval stages of some are parasitic.
	Caligoida (400)	Fresh and salt. Largely ectoparasitic. *Caligus* and *Lepeophtheirus.*
	Lernaepodoida	Fresh and salt. Ectoporasites. *Salmincolar.*
6. *Branchiura* 75	Arguloida	Common ectoparasitic on skin or gill cavity of fresh-water and marine fish. *Argulus* – widely distributed genus. One of favorite hosts – *Amia calva,* the mudfish.
7. *Cirripedia* 800+ Barnacles Adult forms sessile (except for some parasitic forms). Exclusively marine. $100 million yearly cost of fouling to ships.	Thoracica (500+)	*Lepas, Balanus.* Small no. of species widely distributed throughout the world in quantity. Near shore to great depths.
	Acrothoracia (20+)	Boring forms. *Trypetisa.*
	Ascothoracia (20+)	Parasitic on echinoderms and soft corals. *Synagoga.*
	Apoda (1)	Rare. One sp. found by Darwin.
	Rhizocephala (200+)	Parasitic on other crustacea, usually decapods. *Sacculina.*

8. The Subclass Malacostraca of the Class Crustacea must be treated separately because of the large number of species. Many of the species are of great economic importance. There are over 18,000 species of Malacostracans – more than twice as many as all other Crustacea combined. Lobsters, crabs, shrimps, spiny lobsters belong to this interesting subclass. About 66 million pounds of blue crabs, stone crabs, spiny lobsters and shrimp (all Malacostracan Crustacea) were caught in Florida in 1963. The value of these to fishermen was about $17 million.

The body of a malacostracan is composed typically of 19 segments, some of these (the number varies with species) have become fused to form a cephalothorax. Usually there are 13 segments to the cephalothorax and 6 to the abdomen.


Decapoda
Isopoda
Euphausiacea
Amphipoda
Tanaidacea
Cumacea
Copepoda
Ostracoda
Branchiura
Cirripedia
Stomatopoda
Branchiopoda
Mystacocarida
Cephalocarida
Nebaliacea

THE FAMILY TREE OF THE CRUSTACEA
(Simplified from Barnes after Waterman)

(Simplified from: Schmitt, Storer and Usinger, Barnes, Miner)
Subclass – Malacostraca

Superorder	Order
1. Leptostraca	Nebaliacea. Primitive group. Few species. Abdomen 7 somites.
2. Hoplocarida Mantis Shrimp	The Order Stomatopoda. 1-1/2" to over 12" long. Mostly tropical, many caught in shrimp trawls. The 2nd thoracic appendages greatly enlarged and used for catching and killing prey. No commercial value at present.
3. Pericarida	A. Order Mysidacea. 1.5 - 3.0 mm in length. "Opossum Shrimps". Mostly marine. Fresh water form is the food of the Great Lakes Chub. Marine forms food for shad and flounder. B. Order Cumacea. Mostly marine. Small, ± 10 mm. Benthic. C. Order Isopoda. Flattened from top to bottom. About 4000 marine species, many in fresh water. Some terrestrial. Usually lack claws. One species 14" long. D. Order Tanaidaces. Minute, marine, benthic. E. Order Amphipoda. Usually compressed from side to side. Have claws. Wide variety in shape and function of appendages. Mostly scavengers – some parasitic on marine mammals. "Skeleton Shrimp", the caprellids are amphipods. Food for many fish: Herring, mackeral, tuna, flounder.
4. Eucarida Most important group to man. Most of larger malacostracans belong to this group.	A. Order Euphausiacea. Pelagic, shrimp-like, about 1" long. Food for whales. A blue whale may eat 2 or 3 tons per meal (Schmitt). B. Order Decapoda. Shrimp, crayfish, lobsters, crabs. 8500 species – mostly marine. (1) Suborder Natantia (swimming) - Shrimp. (2) Suborder Reptantia (crawling) - Lobster, crayfish, crabs.

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BARNACLES - THEIR HABITS AND LIFE HISTORIES

TO THE TEACHER

This lab is an excellent place to show the application of science in studying a problem of great economic importance. It has been estimated the shipping industry alone spends over $100,000,000.00 per year in correcting damage due to fouling in spite of new hull paints and fouling inhibitors. There is a large barnacle in Chile which is used for food. Many fish with hard mouths feed on barnacles.

TO THE STUDENT

The Classification of Barnacles:

Phylum Arthropoda
Class Crustacea
Subclass Cirripedia
Order Thoracica
Genus *Lepas* or Genus *Balanus*

There are many organisms of interest which either destroy pilings and other wooden structures in salt water by boring or in the case of the barnacle cause damage to ships' hulls by:

1. Increasing the friction between ship and water, thereby increasing fuel consumption or slowing the speed of the ship or both.
2. Reducing the efficiency of underwater acoustic devices, buoys, etc.

The time lost in scraping and repainting hulls is tremendous.

Of the 800 known species of barnacles over 100 species have been classed as fouling organisms.

It is hard to realize that the barnacle is a crustacean. Actually it is a shrimp-like creature which lives on its back with its feet (cirri) sticking up through openings in its shell. The feet are like paddles pushing food into the mouth.

THE PROBLEM UNDER INVESTIGATION

To observe the habits of barnacles.

MATERIALS

Live barnacles
Salt-water aquarium
Strong slit-light
Scalpel and dissecting needles
Dissecting microscope
Petri dishes

PROCEDURE

Barnacles will be found attached to pilings, boards, rocks or almost any object that has been in the salt water for any length of time. Remove the barnacles from their attachment but be careful and do not damage the animal – an old wood chisel is a good instrument for collecting. (1) Put the barnacles in a bucket of salt-water, take to the lab and transfer them to a salt-water aquarium. (2) Strain the water the barnacles were in through a very fine mesh nylon cloth. Wash the cloth into salt water into a petri-dish. Examine under high power of dissecting microscope. Is there anything that looks like the larvae of barnacles, nauplius or cypris? (See sketches of larval barnacles which follow.)

Back to step (1). After the barnacles have become acclimated to their new environment the cirri will begin to move. Put some fine fish food near the cirri and observe how the barnacle feeds. A strong light, properly placed, will assist in this operation. Note that the shell of the barnacle is made up of a series of plates. The plates on top of the barnacle can be closed for protection or opened to let the cirri expand and gather food. The internal anatomy of the barnacle is very difficult to study. Carefully break off the shell from a barnacle and examine under the dissecting microscope (keep the animal under water). The cirri (feet) are obvious but the other parts are difficult to differentiate. There are usually 6 pairs of cirri. See how many there are. The cirri roll up when the shell closes, and unroll when it opens. In a live active specimen see how many times a minute the cirri beat.

Since they hatch and are brooded up to the nauplius stage within the mantle cavity of the barnacle, you may be able to remove some larvae with a pipette. Do this with a specimen which has had part of its shell removed. If found, examine the nauplius under the dissecting microscope.

QUESTIONS ON BARNACLES

1. To what class of Crustacea do barnacles belong?
2. What is a sessile animal?
3. How do barnacles cause damage?
4. Have we developed any methods of reducing the economic loss caused by barnacles?
5. What is the difference between a commensal and a parasitic organism?
6. What is the function of the cirri in barnacles?
7. Can barnacles be used for human consumption?
8. What does the term "indirect development" mean?
9. What are the 2 major types of barnacles?

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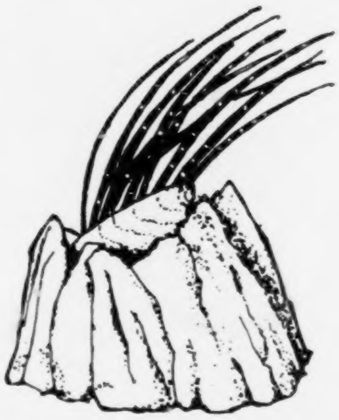
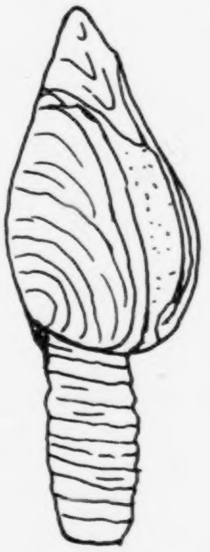
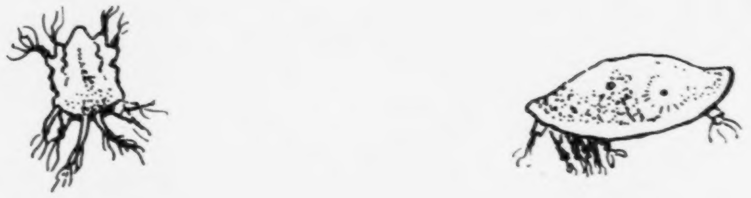


THE ROCK BARNACLE – BALANUS



AN ADULT GOOSE BARNACLE – *LEPAS*



Nauplius

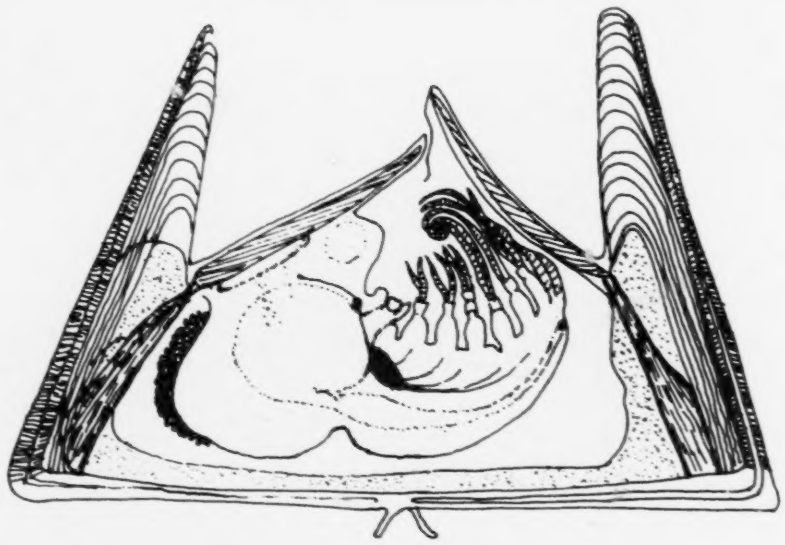
Cypris

STAGES IN THE DEVELOPMENT OF LEPAS

(From Schmitt)

THE INTERNAL ANATOMY OF BALANUS

Part of shell removed to show animal in retracted position



THE FLORIDA BLUE CRAB: *CALLINECTES SAPIDUS* RATHBUN

TO THE TEACHER

A lab exercise of one or two 55 minute periods. The Blue Crab is an ideal laboratory specimen in most areas of Florida because:

1. It is readily available and easy to keep alive.
2. It is large enough so that the various parts can be seen without the aid of a microscope.
3. It is a typical example of the Order of Decapod Crustacea – showing the normal 19 body segments with outstanding examples of specialization of appendages.
4. As a source of food the blue crab is outstanding in flavor. (Obtain a copy of Florida Sea Food Cookery from the State Department of Conservation.)

Classification of the Blue Crab (According to Schmitt)

Phylum Arthropoda
Class Crustacea
Order Decapoda
Family Portunidae
Genus *Callinectes*
Species *sapidus*

The class Crustacea include various species of shrimp, lobsters, crabs, hermit crabs, etc. A comparison of the value of various crustaceans caught in Florida waters or by boats operating from Florida ports might be of interest – in 1963 about $18 million for shrimp, spiny lobster (Florida lobster) over $1 million, blue crabs $1.25 million.

A brief description of the distribution, external anatomy, and reproduction of the blue crab follows. If more details are wanted, see the bibliography. Blue crabs are found along the entire Atlantic Coast and Gulf of Mexico.

EXTERNAL ANATOMY

The body of the crab that is seen from above is called the *cephalothorax* – meaning that the head and thorax have been joined together. The abdomen is small and is folded under the body of the crab. The abdomen of the male is small compared to that of the female. The male abdomen is in the form of an inverted "T". The female abdomen is broadly triangular. Beneath the abdomen of the female are the swimmerets which hold the egg mass.

REPRODUCTION

The female blue crab usually spawns twice during its adult life (1-2 years). Mating occurs only once and that is when she is in the soft stage of her last moult before maturity. During mating the male injects sperm into the female seminal vesicles. About 2 million eggs are produced which are attached in a large egg mass under the abdomen of the female. After hatching, the crab passes through a number of stages. Some students may see these microscopic forms in a marine water sample. See sketches for help in identification.

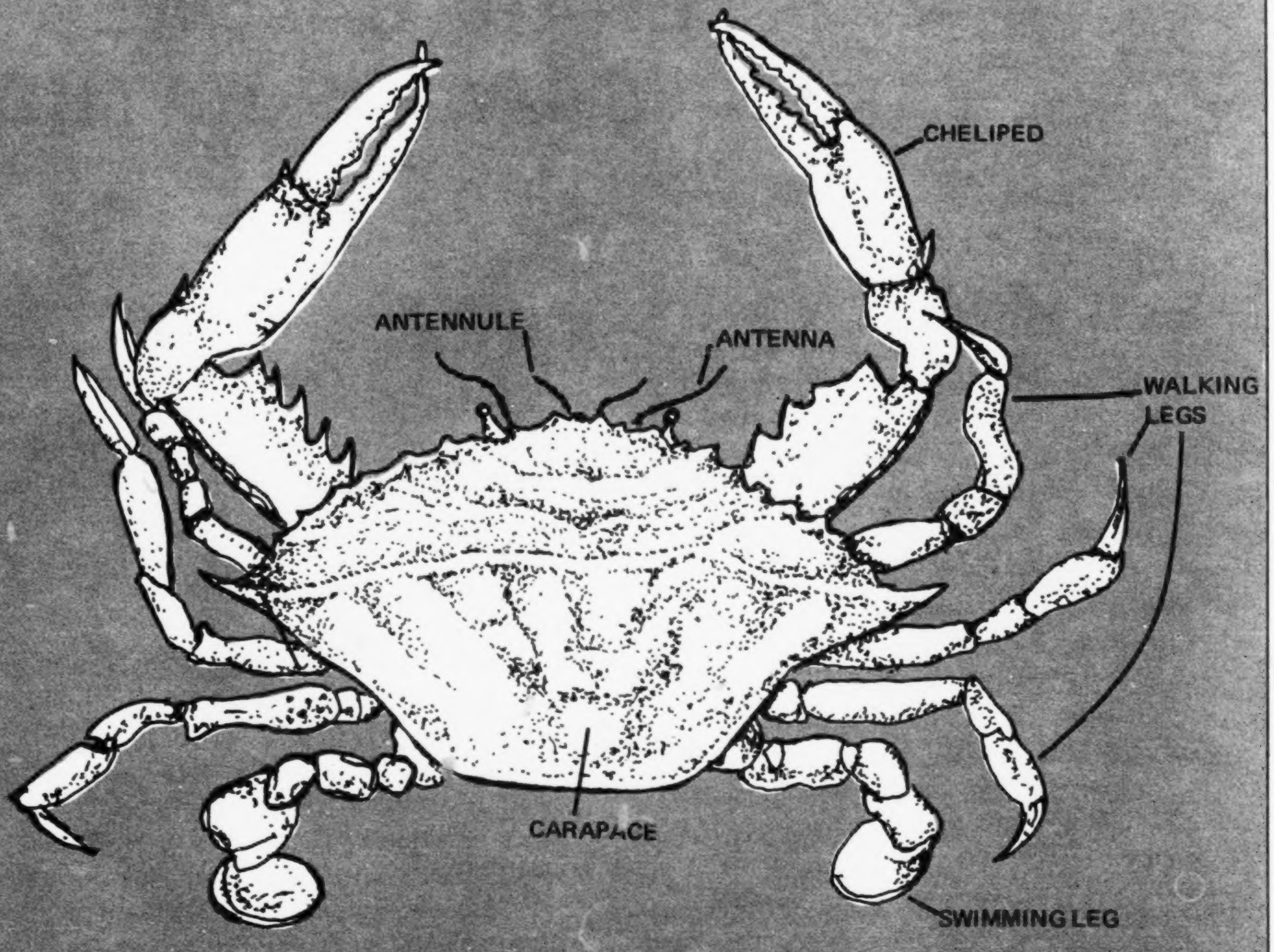
MOULTING

As the blue crab is completely covered with a hard shell, it cannot increase in size without shedding this shell and growing a new and larger shell. This process is called moulting. After the old shell is shed the crab shell is soft for about two days. The crab is a table delicacy during this period. The new carapace may be an inch wider than the old.

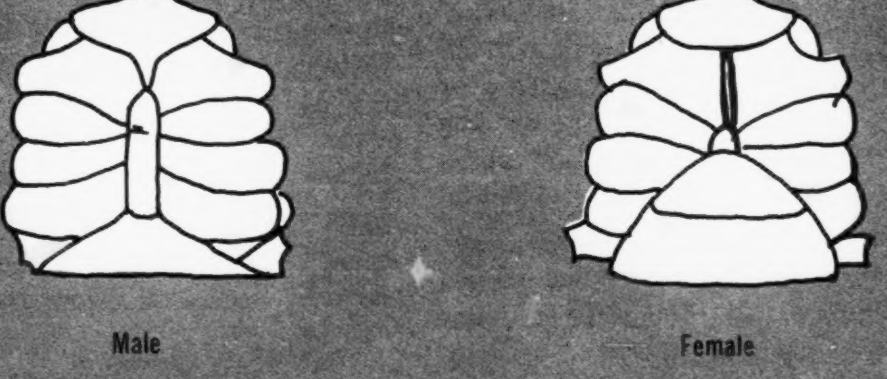
Another characteristic of the blue crab (and crustaceans in general) is their ability to grow a new leg if one has been lost to an enemy. Perhaps crabs with new legs forming will be found.

TO THE STUDENT

In this lab, studied first will be the living crab in which the relationship of structure of appendages is beautifully


CHELIPED
ANTENNULE
ANTENNA
WALKING LEGS
CARAPACE
SWIMMING LEG

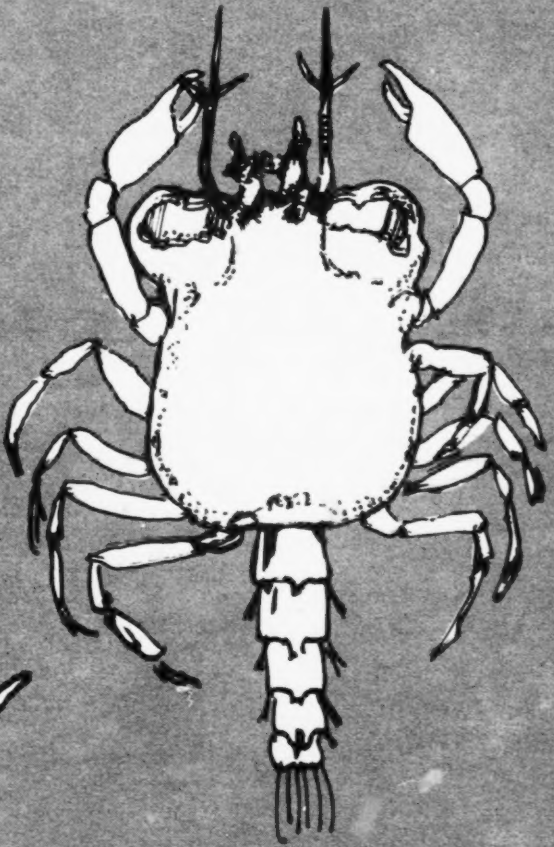
BLUE CRAB—DORSAL VIEW

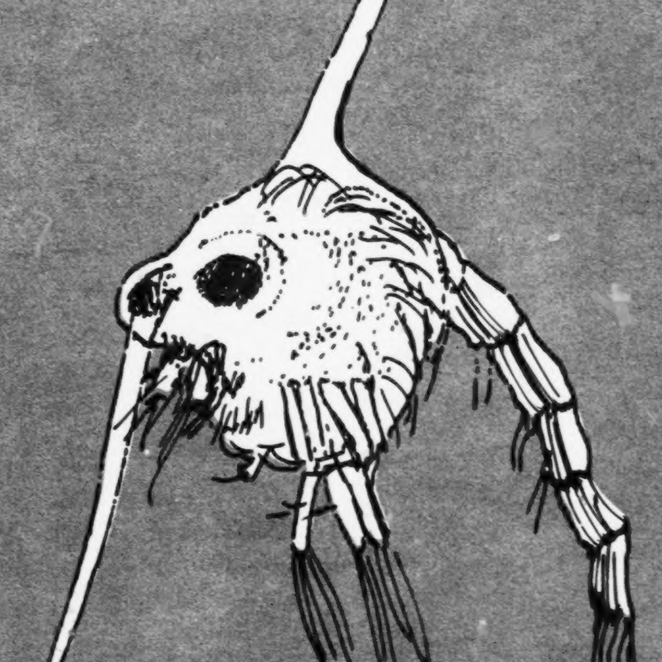

Male

Female

BLUE CRAB—VIEWS OF ABDOMEN

Megalops larva of Blue Crab
Stays in this stage 6-20 days





Zoea larva of Blue Crab
Stays in this stage 31-49 days

The first true crab stage is about 0.1 inch wide. At first, moulting occurs every 3-4 days, then less often until the crab reaches maturity at about 1 year.

related to its various functional needs: feeding, protection, reproduction, swimming, walking, burrowing, feeling, smelling.

MATERIALS LIST

The materials needed are simple. For each student:

1 dissecting pan
2 dissecting needles
1 pair small scissors
1 scalpel
Adult crabs, male and female if possible. The abdomen of the male is the shape of an inverted "T", the female, broadly triangular. Sketch shows both male and female – page 95.

PROCEDURE

1. In an aquarium containing a live crab observe the following:

   a. How does the crab move?

   b. In swimming – are any set of appendages used more than others?

   c. In walking – what appendages are used?

   d. How does it catch live food? (Put a small minnow in the aquarium to see this)

   e. What appendages are used in getting food into the mouth?

   f. Burrowing in sand. Note: See if any of the crabs are forming a new appendage.

   g. What are the crab's defensive weapons? The blue crab has the ability to throw off any of the five pairs of legs. This occurs if a limb is caught by an enemy and the process is called autotomy.

   h. See if you can observe where water enters the gill chamber.

2. In a crab which has been preserved in alcohol or formaldehyde use the line drawing to identify the following:

   a. Antennae and antennules.

   b. The eyes on stalks.

   c. The complex appendages around the mouth.

   d. The heavy paid of chelipeds (pincers or claws) made of the manus, the carpus, and the merus.

   e. Three pairs of walking legs.

   f. The last pair of legs which have their distal segment joint broad and flattened for swimming.

   g. Lift up the abdomen and observe the feather-like appendages (swimmerets).

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SHRIMP-THEIR BEHAVIOR, ANATOMY AND COMMERCIAL IMPORTANCE

INTRODUCTION

Decapoda, of the class Crustacea, the suborder Natantia (the term means free-swimming), includes all shrimp. In Florida, the shrimping industry provides the greatest single source of income to fishermen. In 1967, about 40 million pounds of shrimp were caught with a total value of $13 million. The following chart is based on statistics prepared by Johnson (1970).

Florida Shrimp Catch (heads on) by
area and species 1967

Million of Pounds	Million of Dollars	Area	
4.9	2.5	East Coast	White Shrimp, *Penaeus setiferus* Brown Shrimp, *Penaeus aztecus*
14.3	6.06	Tortugas	Pink Shrimp, *Penaeus duorarum*
3.8	2.1	Campeche	Mostly pink, *Penaeus duorarum*
4.1	1.7	Upper West Coast	Mostly white, some pink, brown and sea bobs, *Xiphopeneus kroyeri*
.7	.32	Central West Coast	
.41	.19	Caribbean	
32.5	12.9		

The above figures are approximate and are used to show the relative value of the various species of shrimp. Several other species of shrimp other than the pink are of value from the Tortugas area. The development of the Tortugas and Campeche pink shrimp industry is a fascinating story but too long to discuss here. The white shrimp (*Penaeus setiferus*) was formerly first in economic importance – now it is the pink shrimp (*Penaeus duorarum*).

TO THE TEACHER

The comparative anatomy of the Crustacea is a most interesting topic. In the Exercise "Introduction to the Marine Arthropods: The Class Crustacea", the general concept of body segmentation and the specialization of appendages in the Crustacea is discussed. The labs on the blue crab, the spiny lobster, and this one on the shrimp can be combined to form a unit extending over several days, or any one lab can be used to advantage. The choice depends on the time available and the geographical location of your school.

TO THE STUDENT

This lab can be divided into two parts. The first is the study of shrimp – living and dead, and the second part should be the study of the habits of shrimp in the ocean. A very good report can be made about the discovery of the Tortugas shrimp beds. This is an example illustrating the fact that a knowledge of the behavior of the shrimp (vertical migration) created a new multi-million dollar industry for Florida. The study of shrimp in the laboratory is a good beginning, but the best place to study shrimp or any other animal is in its own natural environment.

THE PROBLEM UNDER INVESTIGATION

To study the external anatomy and general behavior of any species of shrimp – living and dead.

MATERIALS

1. live and preserved shrimp for the class. If no other shrimp are available, use frozen "heads-on" bait shrimp.
2. dissecting pans
3. petri dishes – cover needed when using live specimens
4. dissecting microscopes
5. dissecting needles
6. stain – carmine red

PROCEDURE

Place a dead shrimp in dissecting pan. Using the dissecting microscope, observe carefully and make a sketch of the following external characteristics: (Internal anatomy is too difficult except in very large Crustaceans).

1. General body shape showing general division into cephalo thorax (head-thorax) and abdomen.
2. Be sure to show rostrum, the sharp spine sticking forward from the head.
3. Antennae and antennules
4. Eyes
5. Legs – show how some legs have special adaptations on last joint.
6. Swimmerets – little swimming feet on abdomen.
7. Tail piece – composed of uropods and telson.

In salt water aquarium containing live shrimp observe the following:

1. What is the normal method of shrimp locomotion?
2. How does the shrimp move when it is in danger?
3. Can the eyes move or are they fixed in one position? Can the eyes move independently of each other?
4. What appendages are used in catching and biting food?
5. What offensive and defensive weapons does the shrimp possess?
6. What behavioral patterns have you observed?

Place living shrimp in sea water in covered petri dish. Use the low power of dissecting microscope.

1. Observe motion of various appendages.
2. Place some carmine red stain in the water with the shrimp. What water currents do you see around or through the shrimp?

CONCLUSIONS FOR TEACHER

This lab should help to show students the economic value of the application of scientific knowledge. It also illustrates the value of increasing our ability to observe seemingly simple phenomena.

The attached bibliography is specialized. More general references may be found in the labs about "The Crustacea" and "Taxonomy of Marine Animals".

REFERENCES

The Florida State Board of Conservation is concerned with all aspects of salt-water fishing. Among their most important contributions are various publications concerning the results of research about commercial shrimping. The following is a partial list only. Anyone who may need additional information should contact the Board of Conservation.

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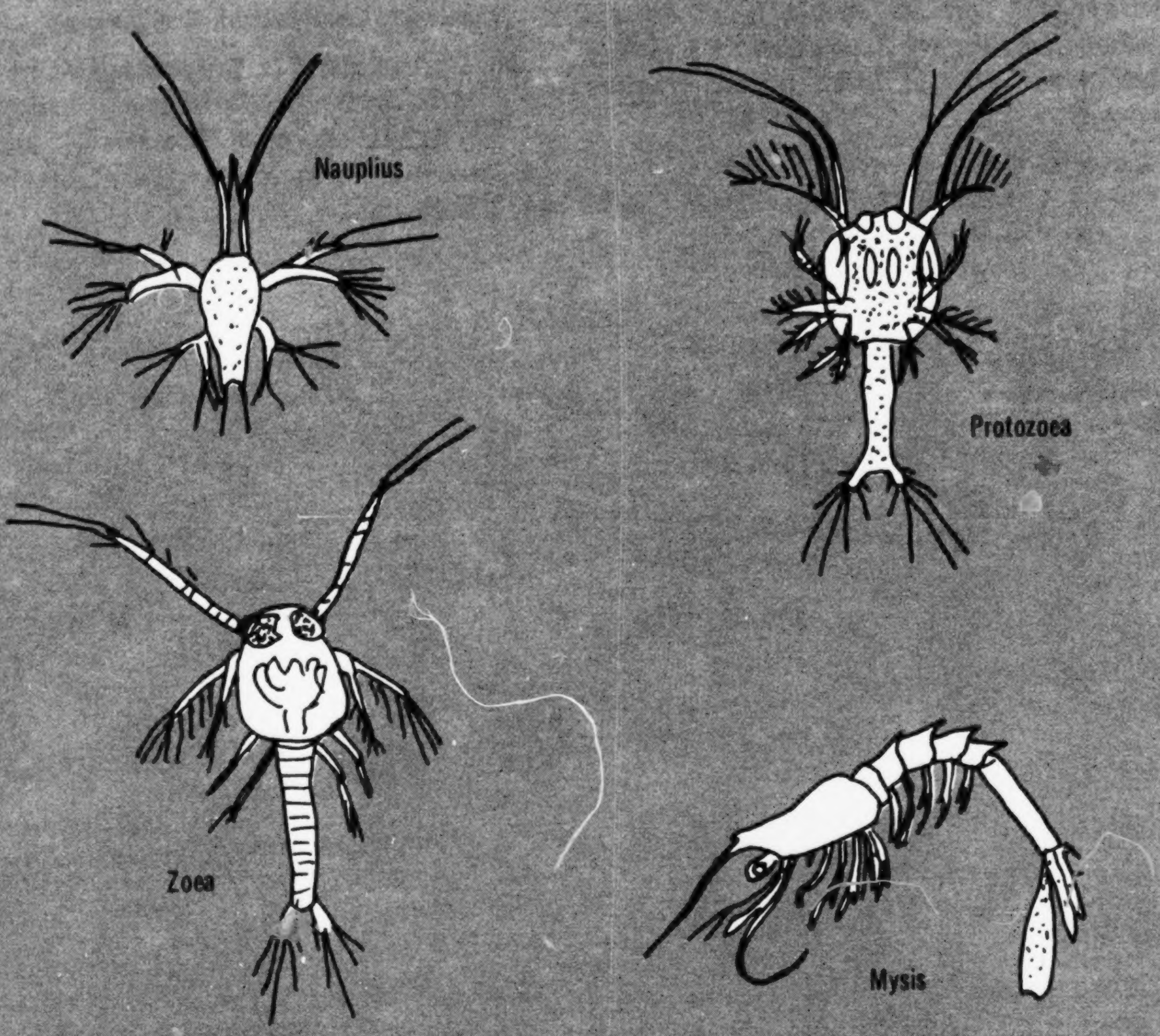
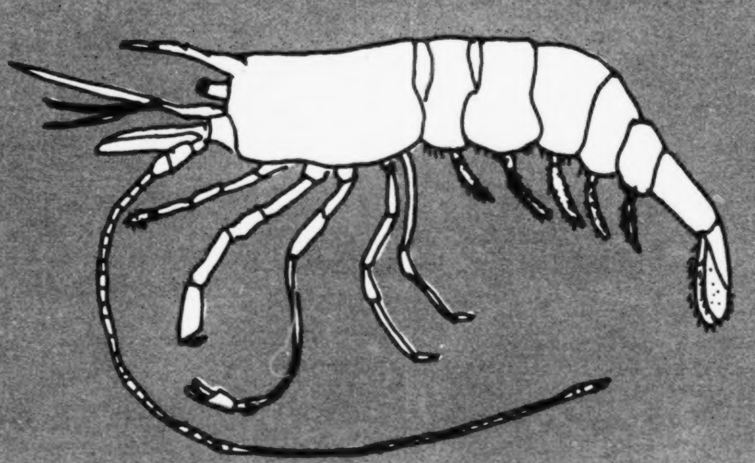
STAGES IN THE DEVELOPMENT OF THE SHRIMP
From Hegner, 1917, College Zoology Simplified


Nauplius
Protozoea
Zoea
Mysis

THE ADULT WHITE SHRIMP—*PENAEUS SETIFERUS* (Linnaeus)
(Simplified from Miner)



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STATISTICAL METHODS

It is strongly suggested that this section, if applicable to the teaching situation, should be presented as early in the semester as possible since many of the laboratory and field exercises call for some degree of statistical analysis. If copies are made for each student, less than one week's time is required to present this unit.

Being able to organize large amounts of data into meaningful terms is a difficult task for the average junior or senior high school student. Yet, an increasing number of laboratory and field activities stress, and frequently result in, an overwhelming pile of quantitative information. For this reason, a basic understanding of statistical methods should become an important part of the secondary science curriculum.

Even a superficial knowledge of statistics, as included in this section, will prove to be a definite asset to students who are in need of a tool to relieve the tedious burden of recording, classifying and interpreting data. It provides them with a step-by-step method of gleaning order and meaning from a mountain of numbers.

A *fictitious* case study is employed to illustrate how statistics are used in fishery biology and, at the same time, to teach students a sequential procedure to follow whenever a statistical treatment of information is needed.

CASE:

Fishery biologists are continually searching for new methods to improve present yields of proteins from the sea to satisfy the demands of an increasing national and world population. One virtually untapped resource, as far as the United States is concerned, is the huge schools of squid reported off our continental shelves.

Japan leads the world's squid fisheries with more than 600,000 tons reported (Walford, 1958). During this same period (1952-53) North American fishermen reported landing about 6,500 tons.

Lane (1960) stated,"... it is probably true to say that the annual catch of cephalopods throughout the world today is in the region of one million tons, or about one pound for every man, woman, and child on earth."

Lane continues, "... In fact the cephalopod population could probably stand several times the present amount of fishing."

Fishery biologists undertook a survey off the Florida east coast to determine the abundance and size distribution of the common squid in that area. The data presented below was the result of one sampling at a depth of 25 meters:

size (in inches)	*number*	*size (in inches)*	*number*
2	1	20	10
3	3	22	14
5	1	23	10
6	4	25	10
7	2	26	5
8	9	27	10
9	1	28	8
10	4	29	10
11	10	30	2
13	8	31	3
16	3	32	4
17	4	35	2
19	12		

At first glance the above data may appear to have limited value. However, as biologists proceed to transfer this information to statistical form its meaning and importance will begin to emerge.

PROCEDURE

Procedures 1-9 refer to this statistical table . . . column by column.

A	B	C	D	E	F	G
i=5	X	f	fX	Δx	Δx^2	$f\Delta x^2$
31-35	33	9	297	+13	169	1521
26-30	28	35	980	+ 8	64	2240
21-25	23	34	782	+ 3	9	306
16-20	18	29	522	- 2	4	116
11-15	13	18	234	- 7	49	882
6-10	8	20	160	-12	144	2880
1- 5	3	5	15	-17	289	1445
		N=150	$\Sigma fX=2990$			$\Sigma f\Delta x^2=9390$

mean (M) = $\Sigma\frac{fX}{N} = \frac{2990}{150} =$ 19.9 or 20 inches

Standard Deviation (SD or σ) =

$$\sqrt{\Sigma\frac{f\Delta x^2}{N}} = \sqrt{\frac{9390}{150}} = \sqrt{62.6} = \pm 7.9 \text{ or } \pm 8$$

1. *Determining Size Intervals or Sets:* The size range of the squid as presented in the table is considerably drawn-out and space-consuming. This disadvantage is quickly eliminated by placing the individual sizes into sets with intervals of 5. (column A)

2. *Determining Midpoint of Intervals:* The middle size in each set. Ranges within sets are generally odd-numbered so that mid-points can be determined with ease, eliminating the involvement of fractions. (column B) Midpoint-X

3. *Frequency:* the number of squid which fall into each set. (column C) N = the sum of f (Σf)

4. *Frequency x Midpoint:* gives the approximate total of scores (inches in this case) in each set (column D). The sum of the frequency x midpoint is expressed as (Σfx).

5. *Determination of the Mean:* using the formula, M = $\Sigma\frac{fX}{N}$, the average size of the squid can be obtained. Specifically, the mean in this case refers to the average arithmetical size of squid (expressed in inches) of that particular sample and at that particular depth.

6. *Determination of Delta* (Δ) *x:* refers to the degree of variability, expressed as plus or minus of the midpoint from the mean. The formula for arriving at Δx is:
$\Delta x = X - M$

Delta x is an intermediate step in determining standard deviation. (column E)

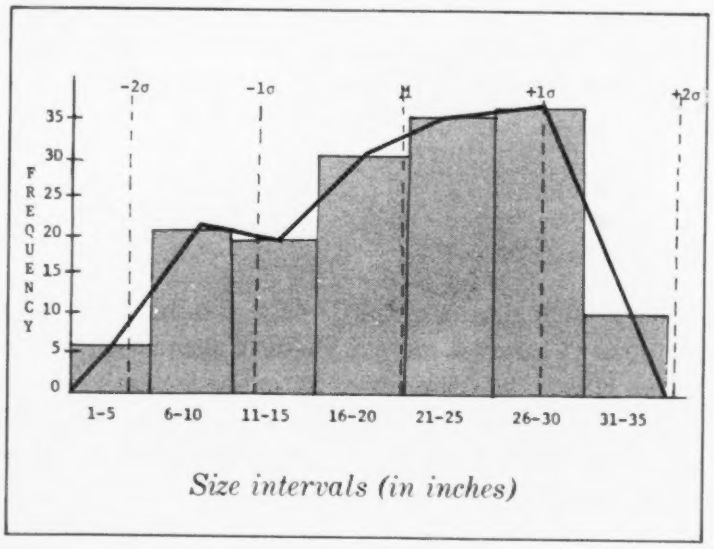
7. *Determination of Delta X^2:* an intermediate step in arriving at SD. Each Δx value is first squared before continuing. (column F)

8. *Determination of Frequency times Δx^2* : This value is obtained by multiplying the frequency within each set with the Δx^2 figure. The sum of $f\Delta x^2$ is used to determine standard deviation. (column G)

9. *Determination of Standard Deviation:* two values are needed to arrive at SD: $f\Delta x^2$ and N. Study the formula above.

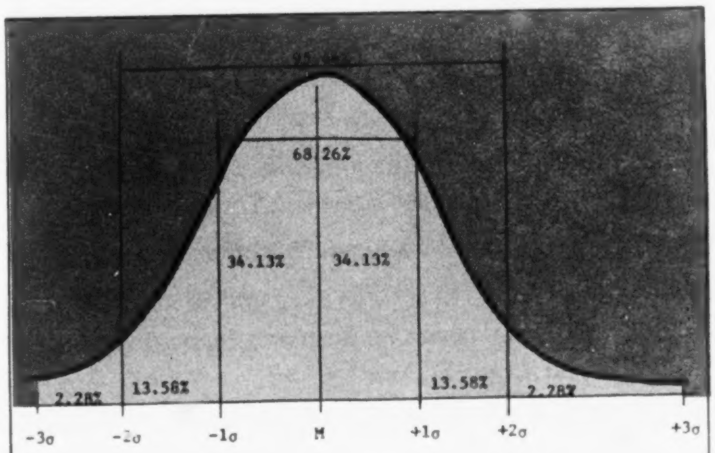
The SD±8 refers to the statistically accepted, average deviation from the mean size of squid (20 inches). The fishery biologists, then, can expect a larger proportion of squid in future samples (at a depth of 25 meters, and assuming that all other factors are constant) to fall within the 12-28 inch size range. True? Fiscuss the degree of validity of this statement.

10. *Plotting the Distribution of the Sample:* The tendency of squid in this sample (and, possibly, in future hauls at 25 meters) to fall within the 12-28 inch size range can best be seen and understood if plotted on a histogram and superimposing a frequency polygon to clarify the resulting distribution of sizes:

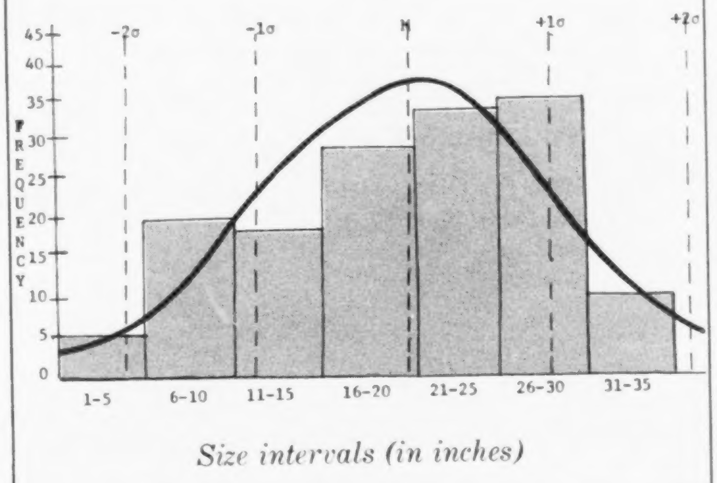


The Size Distribution of 150 Squid Caught at 25 meters Depth off the Florida Coast, September, 1967.

11. *Comparison of Sample Distribution with a Normal Distribution Curve:* The normal distribution curve is a theoretical or ideal picture of a distribution based upon random sampling. Its use is important in statistics because it serves as a standard with which actual distributions can be compared.



Theoretical Distribution of a Random Sample



A Comparison of the Theoretical Distribution Curve with the Actual Size Distribution Histogram of Squid Caught at 25 Meters Depth off the Florida Coast.

What does the above graph show? Discuss.

The plotting of the Theoretical Distribution Curve *cannot* be achieved by guessing or by merely super-imposing a bell-shaped curve on the graph. Rather, the TDC must be carefully computed for each case.

a) *Determining the Height of the TDC:*

$$y = \frac{N}{2.5\,(\sigma i)}$$

y = height of the TDC
N = total sample
2.5 = constant
(σi) = the number of intervals between the actual mean and one SD.

In this sample, one SD is 1.6 (1 3/5) intervals from the mean. Therefore,

$$y = \frac{N}{2.5\,(\sigma i)} = \frac{150}{2.5\,(1.6)} = 37.5$$

Using the y axis (vertical) as a guide, mark the height of the TDC on the actual mean line.

b) *Determining the Height of the TDC Above and Below the Mean:* Once the height of the TDC on the mean line has been calculated, the remainder of the curve can be computed as follows:

Height of the CURVE above and below the mean	1/2 SD=88.3% of y at 1 SD=60.7% of y 1 1/2 SD=32.5% of y 2 SD=13.5% of y

For this sample, then, the relative heights of the TDC, above and below the mean, were:

1/2 SD=33.11
1 SD=22.76
1 1/2 SD=12.18
2 SD=5.05

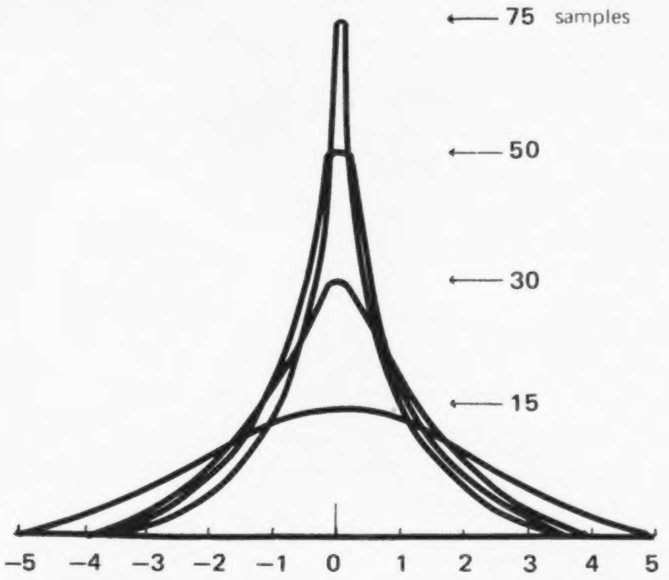
Using the y axis as a guide, mark the heights of the TDC at the designated points. Carefully connect all of the points. What difficulty arises when drawing in the top of the curve?

12. Two of the most important considerations in collecting data are the sampling technique and the sample size. The reliability of statistical analysis depends largely on the limitations of the single bit of datum, but also on the number of tests performed.

All other things being equal, the Gaussian distribution curve will appear normal regardless of the number of tests performed. The average value of a group of tests will be closer to the mean than will the value of a single test.

The larger the sample the closer the average will agree with the mean or, the larger the sample the smaller the standard deviation (σ).

The diagram illustrates the tendency toward the mean as the sample size increases. The distribution retains the normal "bell-shaped curve", but its standard deviation decreases as the *square root* of the number of tests in the sample.



For general biological procedures, a minimum of 50 tests is considered necessary to obtain a valid mean value and workable standard deviation. Below 50 tests the standard deviation increase is fairly large while from 50 to 75 tests the decrease is relatively small.

CONSIDERATIONS AND LIMITATIONS IN USING STATISTICAL METHODS

1. Size of the sample.
2. Specific area in which sampling occurred.
3. Defining conditions under which sampling occurred. (Season, temperature salinity, type of sampling device, etc.)
4. How accurate is the data collected?

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A STATISTICAL ANALYSIS OF A FIDDLER CRAB COLONY

TO THE TEACHER

Thus study was written with the assumption that the required samples of crabs will be provided for the student. The sampling method described in the student section is *fictitious* and *obviously exaggerated* to create a need for later questioning. It would be easy to alter this plan and involve students in the field collecting phase. Allow them to devise their own technique for random sampling of a local colony. Tide (low tide is the best time), size of colony and degree of sampling above and inside burrows are pertinent considerations.

The most important outcome of this exercise should be: a) An awareness of the scientific value and limitations of random sampling. b) Practical experience in applying this technique. c) Statistical methods to an actual biological problem.

TO THE STUDENT

Fiddler crabs are social arthropods. They are found in colonies ranging from less than 100 to many thousands.

Three species of fiddler crabs, from the genus *Uca,* are native to Florida and occur along both the Atlantic and Gulf coasts. Since one claw is greatly oversized, males of the genus are easy to identify. They are frequently observed waving this huge appendage. The outstanding behavioral characteristic of fiddler crabs is the rhythmic elevation and lowering of the oversized claw. The significance of this gesture has been described by various workers as non-sexually territorial, sexually territorial, a sex attractant, a challenge to other males and as various combinations of all these possibilities. (Crane, 1957)

PURPOSE

To statistically analyze the fiddler crab colony at (location) on the basis of a random sample.

MATERIALS

small aquarium
metric ruler
balance
beach sand
saline water (less than 20 ppt)

PROCEDURE

1. Each class will be responsible for the well-being and measurement of 50 crabs.
2. Observe and measure each of the following characteristics:
    a. sex
    b. length of chela (the larger claw if a male; either claw if a female) in mm
    c. mass of crab in grams
3. Record your data for each specimen on the board as follows:
4. Return the crabs to the storage tank . . . alive!

STATISTICAL ANALYSIS

On the basis of class totals calculate and graph, if required:

1. Distribution of sex in the colony.
2. Mean length and standard deviation (SD) of male and female chelae.
3. Mean mass and SD of males and females.
4. Frequency distribution of mass and length.
5. Number of right-clawed and left-clawed males. What percentage of the males were right-clawed?

MALE			FEMALE		
specimen #	length of chela	mass	specimen #	length of chela	mass

QUESTIONS FOR CONSIDERATION

1. What is your opinion of the accuracy of the method described to sample the colony? If needed, how could you improve upon it?

2. What are some other possible limitations of this study?

3. How does the theoretical distribution of sex (50-50?) compare with the actual distribution?

4. Is there any correlation between:
   a. sex and mass?
   b. lengths and masses?

5. On the basis of your statistical analysis and after carefully considering the limitations of this study, what projection(s) can be made concerning the sex distribution, mass and chelae-length characteristics of the colony under study?

6. Aside from studying sex distribution, mass-length relationships, which other characteristics of fiddler crabs could be analyzed?

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THE FLORIDA SPINY LOBSTER *PANULIRUS ARGUS* [dry lab]

TO THE TEACHER

DO NOT COLLECT undersized specimens at any time. Consult your local conservation officer.

TO THE STUDENT

A lab so that you may become familiar with the life history and commercial importance of our Florida lobster.

TAXONOMY: (the Spiny lobster and relatives)

Phylum Arthropoda
Class Crustacea
Subclass Malacostraca
Order Decapoda
Suborder Reptantia
Tribe Palinura
Genus *Panulirus*, species *argus*

The lobster-*Homarus americanus*, (the Northern Lobster) belongs to the Tribe Astacura. The outline sketches with this exercise will give an idea of the difference between the spiny lobster and the northern lobster.

Species of spiny lobster other than *P. argus*, are: *Panulirus guttatus* (southern waters of Florida), occasionally found in large numbers; *P. laevicauda*, few in extreme South Florida; *Justitia longimana*, are deep water species with a large claw on one of the first walking legs. Deep water species are of minor commercial importance as the number of shallow water specimens decreases.

There are closed seasons during the mating period and size limits in catching spiny lobsters. The laws are enforced by the Florida Conservation Patrol. Contact them before doing any lobstering. It is suggested that only one large legal size lobster be used for demonstration to the class.

One feature of this lab is to stress the value of applied scientific knowledge to the study of a commercially important species. It is an excellent place to show how near-shore pollution can kill the adult lobsters as well as destroy the breeding places for the young lobsters. The justification for laws regulating the catching of spiny lobster can be shown by a study of the life history of the lobster.

A brief life history of the spiny lobster can begin with the mating which takes place in early Spring. Spawning begins in April and continues until late Summer, depending on geographic location. The females then move to deep water where hatching takes place. The newly hatched larvae – known as phyllosoma (about 1/10" long) become part of the plankton and for 9 months are carried by various current systems. The next larval stage – the post larva – moves in-shore where growth continues until adulthood. See sketch of larvae in this lab.

THE COMMERCIAL IMPORTANCE OF THE SPINY LOBSTER

At the present time it is impractical to raise lobsters. They are caught commercially by one of the following methods:

1. Traps, 3' x 2' x 2', are made of wooden slats 1-1/2" apart. The traps may or may not be baited. They are weighted to keep them on the bottom and marked with a float. Today these floats are usually a plastic bottle painted with a specific color to identify the owner, and to give visual aid in their recovery.
2. Discarded oil drums or ice molds are dropped to the bottom. They are buoyed as above. Both of the above methods take advantage of the fact that lobsters like to hide under objects on the bottom.
3. Hoop nets.
4. Bully nets – used at night with a light.

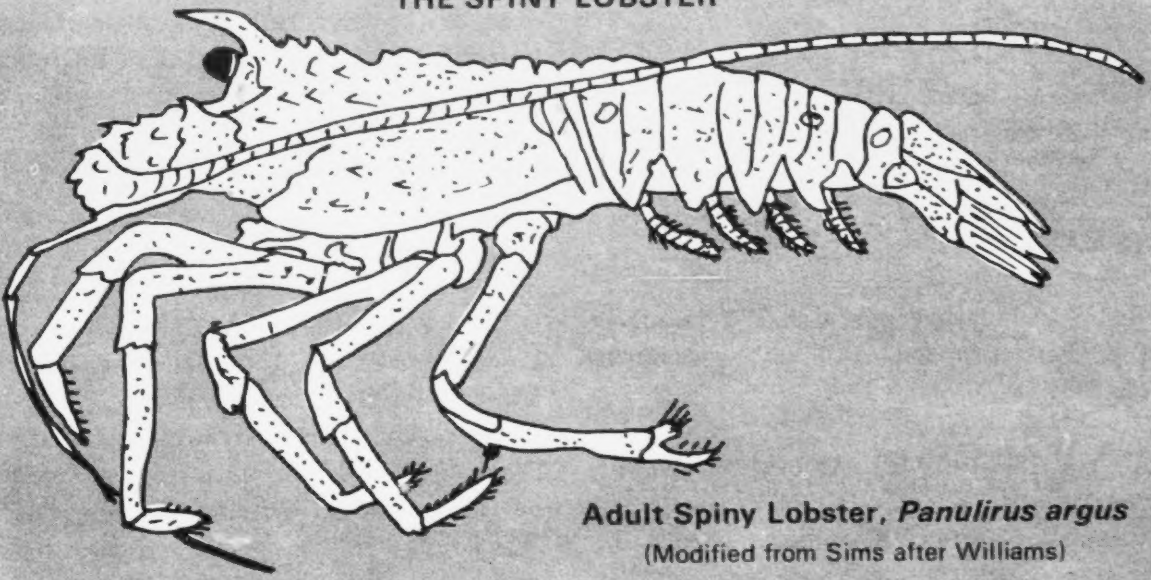
In 1963, in Florida, 3,585,000 pounds of spiny lobster were caught. The value to fishermen was $1,400,000.

The spiny lobster is a scavenger. It is not fast enough to catch many live fish, but will eat snails or anything else available.

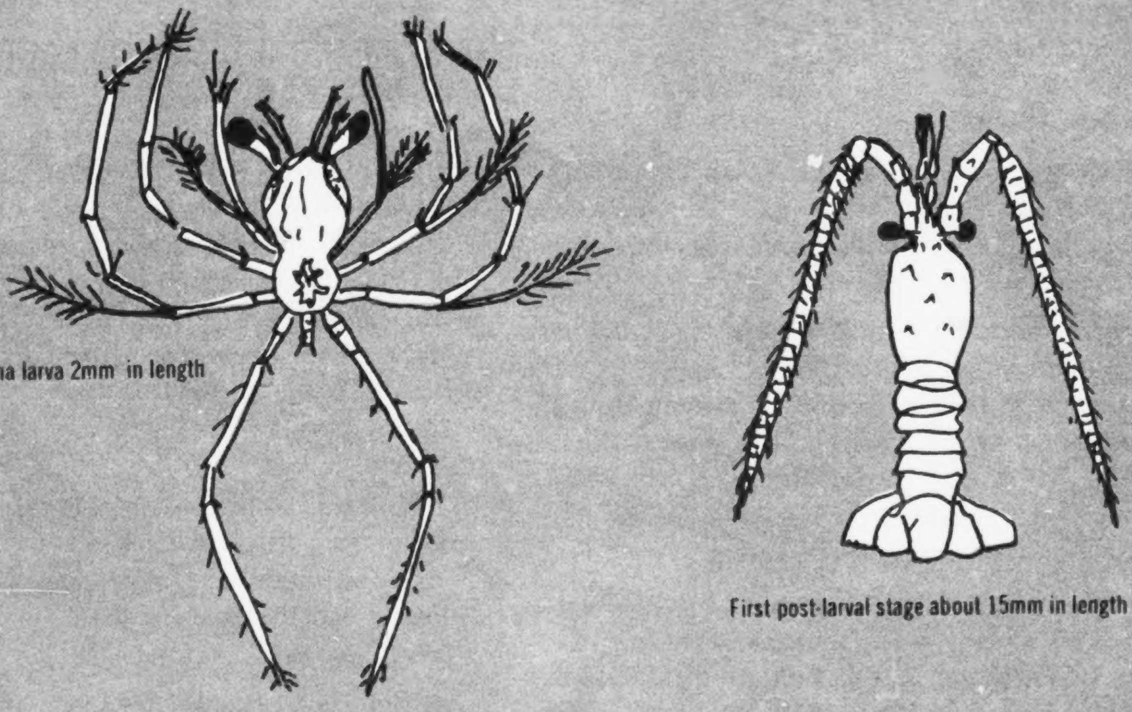
QUESTIONS FOR CONSIDERATION

1. What is the difference in the appendages between the northern lobster and the spiny lobster?
2. Why are there closed seasons on spiny lobsters?
3. What state agency enforces the laws pertaining to lobstering?
4. What are the stages in the development of *Panulirus argus?*
5. What methods are used in catching lobsters in Florida?

THE SPINY LOBSTER



Adult Spiny Lobster, *Panulirus argus*
(Modified from Sims after Williams)

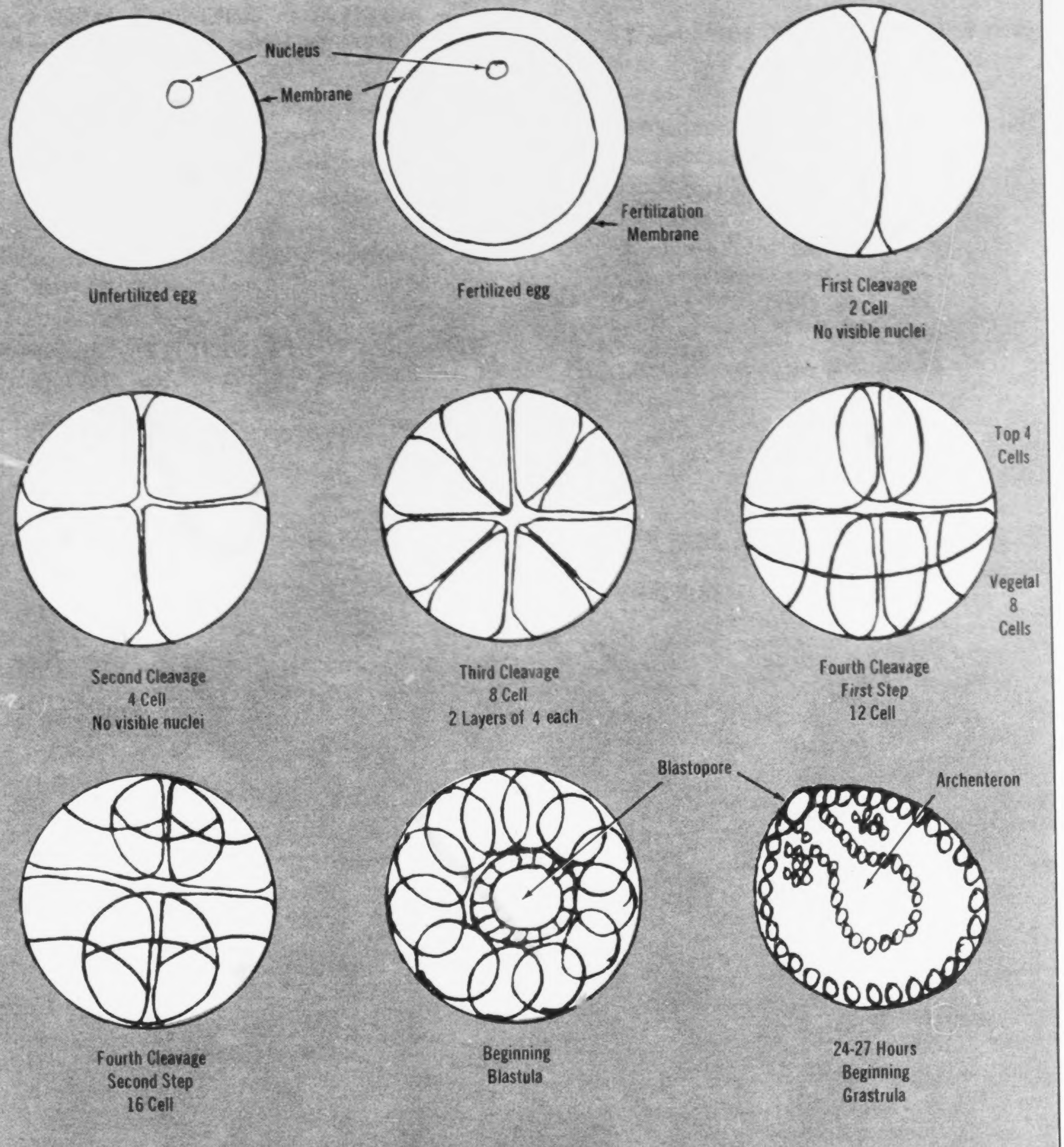


Phyllosoma larva 2mm in length

First post-larval stage about 15mm in length

Larval Stages in the Development of the Spiny Lobster
(After Sims)

CLEAVAGE DIAGRAMS


Nucleus
Membrane
Fertilization Membrane
Top 4 Cells
Vegetal 8 Cells
Blastopore
Archenteron

Unfertilized egg

Fertilized egg

First Cleavage
2 Cell
No visible nuclei

Second Cleavage
4 Cell
No visible nuclei

Third Cleavage
8 Cell
2 Layers of 4 each

Fourth Cleavage
First Step
12 Cell

Fourth Cleavage
Second Step
16 Cell

Beginning
Blastula

24-27 Hours
Beginning
Grastrula

After Hallas, D.J. unpublished—Largo, Fla.

Reprinted by permission of David J. Hallas, author of The Study of Lytechinus Variegatus As A Laboratory Animal for Embryological Exercises in a High School Biology Program. 1965.

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SEA URCHIN FERTILIZATION AND DEVELOPMENT

TO THE TEACHER

Sea urchins present an excellent opportunity to illustrate the process of reproduction. Including: (a) morphological structure of sperm and egg; (b) fusion of gametes; (c) development of the fertilized egg.

Two species of sea urchins are recommended for use as laboratory animals, ***Arbacia punctulata*** and *Lytechinus variegatus.* These are relatively common in occurrence, easy to collect, hardy in the uncrowded aquarium and possess ripe eggs and sperm from September to June (probably best in February, March, and April).

There is no external feature that can be used to separate the sexes in the field; therefore, it is recommended that 10-12 specimens be collected to insure obtaining at least one member of each sex. To insure the collection of mature specimens, select only those specimens with a test (shell) diameter of from 6-8 cm.

The instructor should prepare a fresh supply of eggs and sperm each hour. This demonstrates the method of obtaining gametes to the class and insures and uncontaminated supply.

The urchin is washed in cold running tap water for two or three minutes to remove detritus and eggs or sperm that may have collected on its test, then rinsed in fresh filtered sea water.

Place the animal with its oral side up (inverted from its normal position) and cut the test along the area of its greatest circumference.

The oral or upper portion of the test is now discarded and the aboral (lower portion) is rinsed with filtered sea water.

It is possible to identify the sex in *some* urchins. The female gonad (ovary) is red or orange colored and the male gonad (testes) is white.

Remove the female gonads using a spoon and place them in a petri dish or finger bowl of fresh filtered sea water and allow to stand five to ten minutes. Ripe gonads will immediately begin to eject gametes. The eggs will appear as yellow spheres and sperm as a milky fluid.

Filter the eggs through cheese cloth to remove detritus and pieces of tissue. Place the eggs in 250-500 ml of fresh filtered sea water. They will remain healthy for one to two hours. The eggs will settle to the bottom of the container. They can be removed with a pipette as needed.

The testes can be stored in a petri dish without water in a refrigerator (8°C) for four or five days and remain healthy. Sperm become active when placed in sea water, but die after approximately one hour.

After each urchin has been opened, all instruments (including your hands) must be washed thoroughly. This will prevent contamination of subsequent specimens.

Prepare the sperm and egg suspension for the class by adding one drop of sperm to 1 ml of eggs in 250 ml of fresh filtered sea water. This should be sufficient for 25-30 students.

A second method is to place 1 ml of eggs in a petri dish and add sperm with a toothpick using only the amount of sperm picked up by the end of the toothpick. Stir the solution gently with the toothpick to insure even distribution.

One to two hours are required for completion of this exercise.

TO THE STUDENT

Sea urchins are mentioned in the writings of many of the ancient Greeks and Romans as a food. Urchin eggs are still considered a delicacy in many parts of the world. Some brave souls in the class may wish to taste this particular brand of "caviar".

The ancients, according to Pliny (23-79 A.D.), used sea urchins as medicine. The urchin was ground, spines and all, mixed with a cup of wine or vinegar, and quaffed. In some cases, urchins were burned with snake skins and frogs, the ashes were mixed with vinegar. One cup a day was drunk to improve eyesight.

This exercise illustrates the principles of fertilization and development of the egg.

Sea urchins normally discharge gametes into the sea as the water warms in the spring. This process, external fertilization, is very common among aquatic organisms.

After a single spermatozoan has penetrated the egg, a fertilization cone is formed which may last five minutes. Secondly, a fertilization membrane rises over the cone and covers the entire egg in a few seconds. The egg now elongates and the first cleavage begins, leading to the ultimate formation of the free swimming pluteus.

PURPOSE

To observe the development of a zygote during its early stages of division.

MATERIALS

microscope
sea urchins
petri dishes, watch glasses
slides (plain, concave, or Blister)
cover slips
fresh sea water, filtered
pipette
small pieces of glass tubing
cheese cloth
Vaseline
scissors

PROCEDURE

1. Secure a drop of the sperm and egg suspension. Place on a hanging drop slide (sealed in Vaseline), a concave slide (cover slip sealed in Vaseline), or a Blister Slide.

2. Record observations made under the microscope. Use 10x objective.

3. After completing observations, place the slide on two glass rods in a petri dish, add water to cover 1/2 of the rod but not the slide. This will provide a moist incubation chamber and will possibly allow observations to continue up to 48 hours.

4. Repeat microscopic observations as often as possible during the day.

ORGANIZED DATA RECORDING

1. Measure size of unfertilized egg, sperm, and initial fertilized egg. (See lab: "Measuring with a Microscope")

2. Record the time lapse between the mixing of sperm and egg to formation of:
   a. fertilization cone
   b. fertilization membrane
   c. first cleavage
   d. second cleavage
   e. third cleavage
   f. fourth cleavage
   g. blastula
   h. gastrula
   i. pluteus

3. Record observed physical changes that occur in the stages listed above.

QUESTIONS FOR CONSIDERATION

1. How is development of the sea urchin zygote similar to development in mammals?

2. In what direction does cleavage occur in each step of division?

3. Compare the motility of egg and sperm.

4. How do you explain "parthenogenetic" development in sea urchin eggs?

5. What are the advantages and disadvantages of liberating sperm and eggs into the oceans?

LIMITATIONS AND SOURCES OF ERROR

Use only freshly obtained animals

Fresh sea water

Evaporation of sea water from observation chamber

Clean glassware

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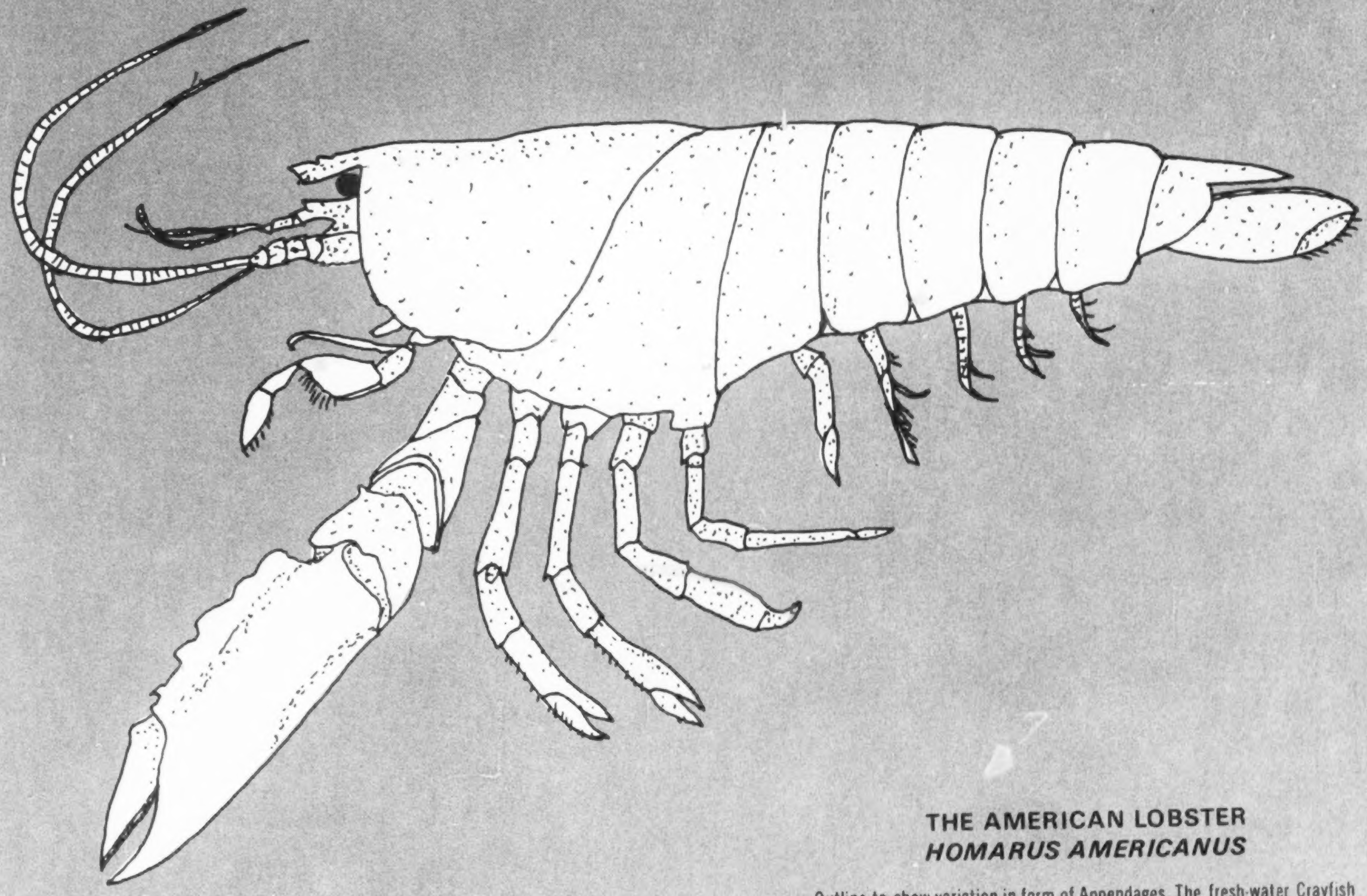


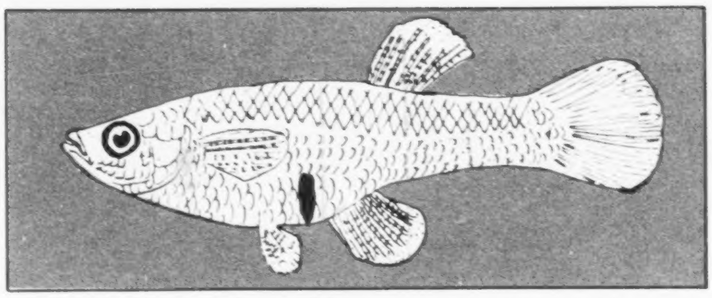
THE AMERICAN LOBSTER
HOMARUS AMERICANUS

Outline to show variation in form of Appendages. The fresh-water Crayfish resembles a miniature lobster.

EMBRYOLOGY OF LIVE BEARING FISHES

INTRODUCTION

Large numbers of brackish water and fresh water fishes utilize the process of internal fertilization by means of the male fish possessing a modified anal fin. Among these ovoviviparous fish, the female retains the eggs in the ovary during the embryonic development period. At birth the fry are ready to care for themselves, indicating that a great amount of development occurs before birth.



female *Gambusia sp.*

TO THE TEACHER

This exercise requires on laboratory period for part A and two laboratory periods for part B.

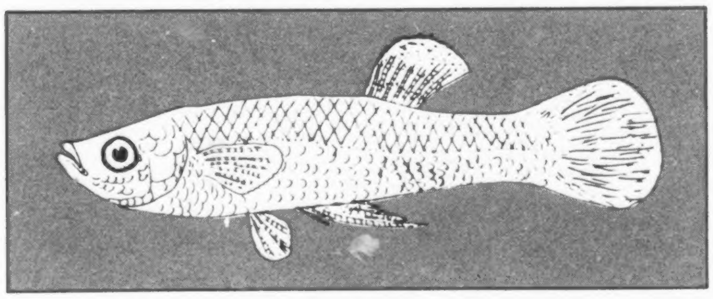
If sufficient student interest is developed, they may wish to explore courting behavior, population counts, or sexual dimorphism.

The fish to be used include: *Gambusia affinis* (Mosquito Fish); *Heterandria formosa* (Dwarf Top Minnow); and *Mollienisia sp.* (Sailfin Molly). These can be obtained by seining fresh to brackish water, ditches, pools, and ponds. If these are not available, *Lebistes reticulatus* (Guppy) will suffice.

Each student must dissect several gravid females in order to obtain a large range of embryonic stages.

The mature gravid female of the *Gambusia sp.* is easily recognized by its swollen abdomen and large dark pigmented spot. (see illustration)

A supply of specimens (5 per student) should be on hand before beginning.



male *Gambusia sp.*

TO THE STUDENT

The mature female *Gambusia sp.* is gravid almost continuously. Secondly, the female of the species is usually larger than the male and more numerous. The female of some fish species such as *Gambusia sp.* or *Lebistes reticulatus* possess a dark pregnancy spot. In choosing your specimens for dissection, select fish with spots of varying size and intensity of pigmentation.

The female Gambusia exhibits superfetation (the possession of more than one set of embryos at a time) by retaining sperm from the initial mating and producing a second set of eggs while the first is developing. These sperm are used to fertilize the second set of eggs during the development of the first set. The development of the second set of eggs is retarded until the birth of fry, then development begins rapidly.

The average time required for the development of the egg until fry are produced is 28 days.

The ovoviviparous fishes have internal fertilization, internal development of the fry, and live birth, but there is no placenta (the organ in most mammals by which the fetus is joined to the uterus and is nourished). The fertilized egg develops without benefit of attachment to its mother; instead it depends on the yolk.

PURPOSE

To examine and determine the varying stages of embryonic development in an egg during its growth.

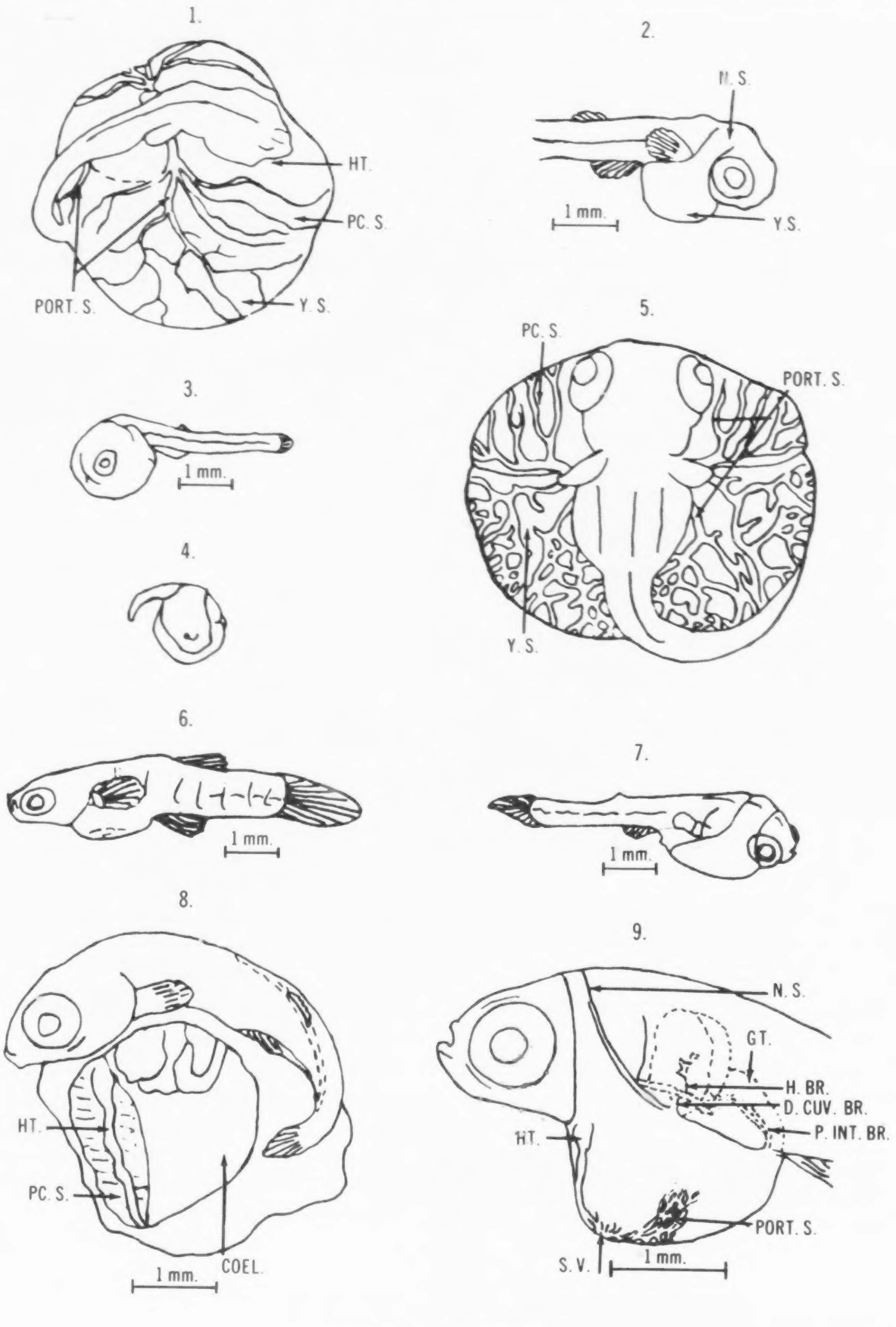

1.
HT.
PC. S.
PORT. S.
Y. S.
2.
N. S.
1 mm.
Y. S.
3.
1 mm.
4.
5.
PC. S.
PORT. S.
Y. S.
6.
1 mm.
7.
1 mm.
8.
HT.
PC. S.
1 mm.
COEL.
9.
N. S.
GT.
H. BR.
D. CUV. BR.
P. INT. BR.
HT.
PORT. S.
S. V.
1 mm.

PLATE OF DRAWINGS

PLATE OF DRAWINGS

Key to abbreviations:

COEL., coelom
D.CUV.BR., branch joining portal system from duct of Cuvier
GT., gut
H.BR., hepatic branch joining portal system
HT., heart
N.S., neck strap
PC.S., pericardial sac
P.INT.BR., posterior intestinal branch joining portal system
PORT.S., portal system of embryo
S.V., sinus venosus
Y.S., yolk sac

Key to description:

1. Seven-day embryo of *Fundulus heteroclitus:* To illustrate spreading of portal system to pericardial sac.
2. Five and five tenths mm embryo of *Gambusia affinis:* To illustrate typical neck strap structure.
3. Three and five tenths mm embryo of *Heterandria formosa.*
4. Very young embryo of *Heterandria formosa.* Head is enveloped by the external and internal folds of pericardial sac.
5. Nine-day embryo of *Fundulus heteroclitus* somewhat flattened dorso-ventrally.
6. Embryo of *Heterandria formosa* at close of the period of gestation. Neck strap has been absorbed and belly sac is shrinking.
7. Five mm embryo of *Heterandria formosa.* Head has pushed through the folds of the pericardial sac leaving a neck strap dorsally.
8. Diagram of a dissection of 5 mm embryo of *Poecilistes sp.* Portal network covering belly sac is not indicated.
9. Diagram of the portal system covering the yolk sac in a 7.5 mm embryo of *Lebistes reticulatus.* Blood emerges from the body of the embryo through branched arteries via the liver, duct of Cuvier, and region just in front of the anus, traverses the portal network and is received in the sinus venosus.

The abbreviations, illustrations and descriptions are adapted from Turner (1940a).

PART A- PRESERVED SPECIMENS

MATERIALS

5 gravid females (preserved)
dissecting needle
small pair of sharp scissors or razor blade
watch glass or petri dish half
dissecting microscope

PROCEDURE

1. Secure preserved specimens and petri dish.
2. Measure the standard length of each specimen.
3. Hold the head between your fingers and cut the head from the body behind the gills.
4. Cut the abdominal wall back to the anal opening, thus opening the stomach cavity.
5. Place under the dissecting scope and remove the white to yellow spheres observed in the viscera. These will be the embryos and eggs.
6. Sort these spheres into groups according to the amount of development observed.

PART B- LIVE SPECIMENS

MATERIALS

live fish
physiological saline (0.7% solution of NaCl in distilled water)
items listed under materials in part A

PROCEDURE

1. Instructions 1-5 are the same as in part A.
2. As the embryos are removed from the viscera they are to be placed in a physiological saline solution and maintained at room temperature for three to four days and observed at regular intervals. Time alive should be recorded.

3. If the embryo is well-developed at the time of dissection, it is possible to place them in an aerated aquarium and observe development to maturity.

ORGANIZED DATA RECORDING

Record the number of eggs and/or embryos removed from each female and the various stages of development for the eggs and embryos. Record the number of males and females in the sample of specimens. Records of the standard length should accompany each female.

QUESTIONS FOR CONSIDERATION

1. These fish do not produce as many eggs as some others. Why?

2. Can you find any correlation between the size of the pregnancy spot and the number of eggs and embryos per female?

3. Why are not all of the eggs and embryos at the same stage of development?

4. What is the function of the placenta in animals?

5. How does the *Gambusia sp.* fry eliminate waste material before birth?

GRAPHIC ANALYSIS

1. Graph standard length against the number of eggs and embryos per female.

2. Graph the development of eggs and embryos at each level of development.

3. Calculate the percentage of females versus males in the total sample.

LIMITATIONS

Each student should have five to ten specimens available to insure a wide range of embryonic development.

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A SHARK STUDY

TO THE TEACHER

In the eyes of the high school student of marine science, the shark provides a stimulation equaled by no other animal of the sea. This laboratory study is constructed so that the class can be involved for four consecutive periods (one week's work). On the other hand, any part can serve by itself. Depending upon time allowed, specimens available and level of interest, any or all of the separate exercises may be expanded.

Basic Outline of Study Sequence:

- I Reading Research
- II External Anatomy
- III Internal Anatomy
- IV Brain Dissection
- Testing (optional)

Local sport and commerical fishermen will help in collecting a supply of embryo sharks. Nearby professional marine laboratories, especially those engaged in routine shark studies, can also provide specimens. Be ready to respond immediately to the call of the volunteer collector, and have storage facilities ready at all times: freezer facilities, capable of storing the embryos, are recommended. The animals can be preserved in ordinary fixative, if there are no freezer facilities.

The embryos of the brown shark [*Carcharhinus milberti*, (Muller and Henle)] reach about 50 cm before birth. Ideally, this laboratory calls for two students per shark.

Collection of live adult specimens by high school students should be *discouraged*. Even the more docile nurse shark *Ginglymostoma cirratum* (Bonnaterre)] will bite if handled improperly. (Gilbert 1964).

TO THE STUDENT

Shark classification [from Perlmutter (1961)]: Class: Chondrichthyes (Elasmobranchs and Chimaeroids)

Subclass: Elasmobranchii (Sharks, Rays, Skates)

Order: Selachii (Modern Sharks)

Suborder: Galeoidea

Family: Carchariidae (Sand Sharks)
Carcharias taurus – sand shark

Family: Isuridae (Mackerel Sharks)
Lamna nasus – mackerel shark, porbeagle
Isurus oxyrinchus – mako, sharp-nosed mackerel shark
Carcharodon carcharias – white shark, man-eater

Family: Cetorhinidae (Basking Sharks)
Cetorhinus maximus – basking shark

Family Alopiidae (Thresher Sharks)
Alopias vulpinus – common thresher shark

Family: Orectolobidae (Carpet Sharks)
Ginglymostoma cirratum – nurse shark

Family: Orectolobidae
Family: Rhincodontidae (Whale Sharks)
Rhincodon typus – whale shark

Family: Scyliorhinidae (Cat Sharks)
Scyliorhinus retifer – chain dogfish
Apristurus profundorum – deep-water cat shark

Family: Pseudotriakidae (False Cat Sharks)
Pseudotriakis microdon – false cat shark

Family: Triakidae (Smooth Dogfishes)
Mustelus canis – smooth dogfish

Family: Carcharhinidae
Galeocerdo cuvier – tiger shark
Paragaleus pectoralis – Paragaleus
Prionace glauca – blue shark
Scoliodon terrae-novae – sharp-nosed shark
Aprinodon isod[illegible] – smooth-tooth shark
Negaprion brevirostris – lemon shark
Carcharhinus falciformis – sickle-shape shark
Carcharhinus leucas – cub, ground, bull shark
Carcharhinus limbatus – spot-fin ground shark, small black-tipped shark
Carcharhinus milberti – brown shark
Carcharhinus obscurus – dusky shark

Family: Sphyrnidae (Hammerhead Sharks)
Sphyrna tibura – shovelhead, bonnet shark
Sphyrna diplana – southern hammerhead shark
Sphyrna zygaena – common hammerhead shark

Suborder: Squaloidea

Family: Squalidae (Spiny Dogfishes)
Squalus acanthias – spiny dogfish
Centroscyllium fabricii – black dogfish
Centroscymnus coelolepis – Portuguese shark

Family: Dalatiidae
Somniosus microcephalus – Greenland shark

Suborder: Squatinoidea

Family: Aquatinidae
Squatina dumeril – angel shark

Sharks are chordates with a skeleton made of cartilage. The gills open to the outside of the body through slits. Fertilization is internal and, in most species, the young are born alive. Sharks are scavengers and most are flesh-eaters. Their senses of smell and hearing are very well-developed. Teeth of the shark occur in series or rows. Tooth replacement occurs throughout the life of the shark.

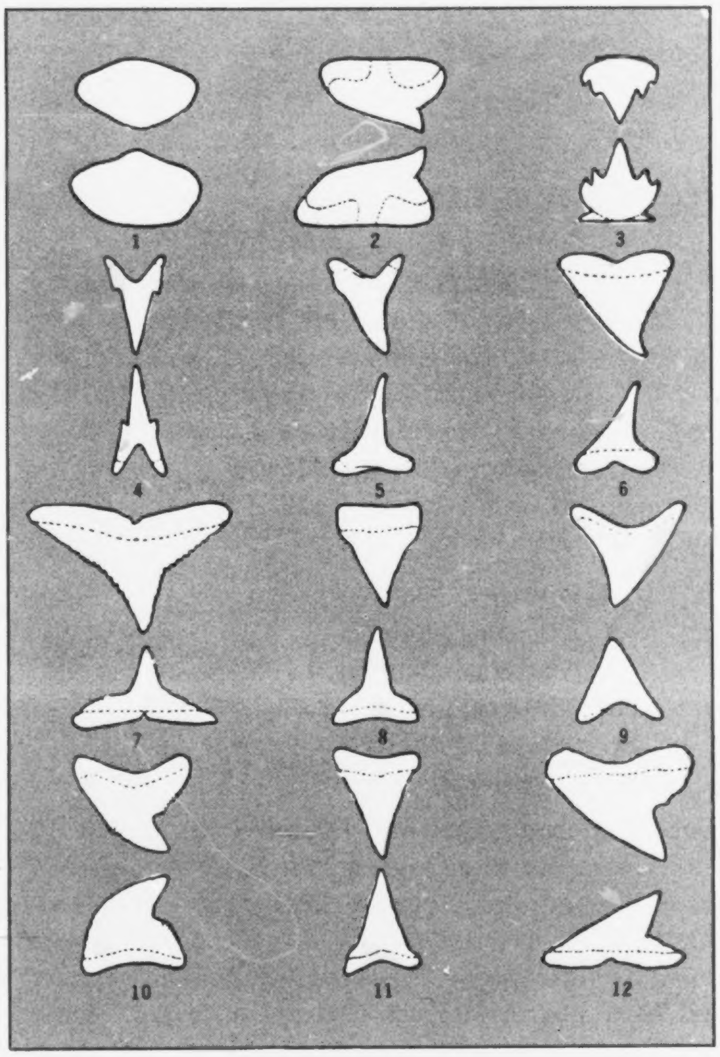
Sharks' attacks upon man occur with greatest frequency between 30°N and 30°S latitudes. Since their actions are so unpredictable, most experienced divers and collectors treat the shark with caution and respect.

> *Species of Sharks Known to be Dangerous to man:* The sharks believed to be of greatest concern to the diver are members of four families. These families are listed according to what is believed to be their order of greatest danger to man, namely:
>
> *Isuridae* – the Mackerel, or Man-eaters
> *Carcharhinidae* – the Requiem Sharks
> *Carchariidae* – the Sand Sharks
> *Sphyrnidae* – the Hammerhead Sharks
>
> Halstead (1959)

The following species are fairly common in Florida waters:

- Blacktip Shark
- Bonnethead Shark
- Brown or Sandbar Shark
- Bull Shark
- Dusky Shark
- Hammerhead Shark
- Lemon Shark
- Nurse Shark
- Tiger Shark

If lab exercises are scheduled on the internal organs, heart and brain, make appropriate anatomical sketches from reading research for future reference.



An upper and lower tooth to the left of the symphasis. 1. Smooth dog shark; 2. Spined dogfish; 3. Nurse shark; 4. Sand shark; 5. Mackerel shark; 6. Blue shark; 7. Spot-fin ground shark; 8. New York ground shark; 9. Thresher shark; 10. Tiger shark; 11. Man-eater shark; 12. Hammerhead shark. (Breder)*

THE PROBLEM

1. Through reading research, to become acquainted with the taxonomy of sharks; to learn about the feeding habits of sharks; and to become familiar with present-day commercial and research interests in sharks.

2. To measure and record the external features of a shark embryo.

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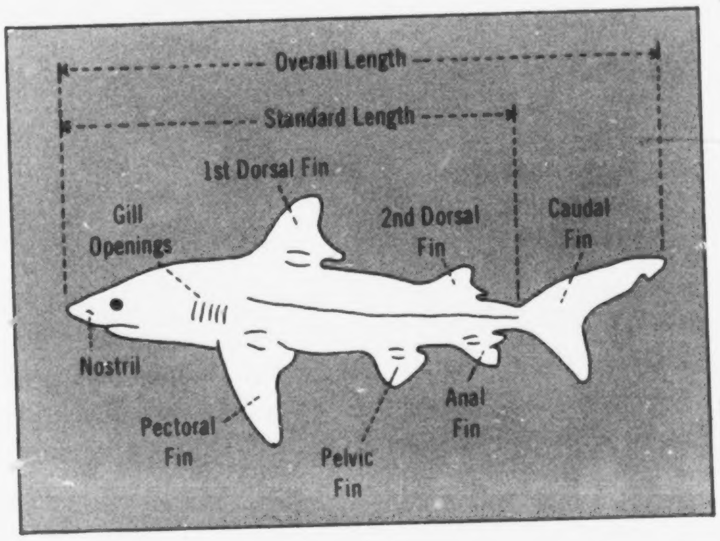
3. To learn about the gross internal anatomical features of a shark embryo.

4. To learn about the gross anatomy of the brain of a shark embryo.



External anatomy of a typical shark.

MATERIALS

shark embryo
dissecting board
newspapers and paper towels
dissecting kit
scales
chart
calipers and ruler
anatomical charts

PROCEDURE

Part I. Reading Research

A. Complete the classification of your specimen:

Phylum: ____________________
Class: ____________________
Subclass: ____________________
Order: ____________________
Suborder: ____________________
Family: ____________________
Scientific Name: ____________________
Genus Species

B. Answer the following questions. State the source(s) of your reference(s).

1. List the physical characteristics which separate the sharks from the true fishes (Class Osteichthyes).
2. What are the feeding habits of sharks?
3. Of what economic importance are sharks?
4. Of what research interest are sharks? Where is research on sharks being carried on in Florida?
5. Under what conditions are sharks most dangerous to man?
6. What other animals are in the same class category as sharks?
7. What species of sharks are found in the coastal waters nearest the school?

Part II. External Observations

Record the following data:

____________________ ____________________
Common Name Scientific Name

Length (overall) ____________________ cm
Length (standard) ____________________ cm
Width: at Head (A-A) ____________________ cm
at Middle (B-B) ____________________ cm
at 2nd Dorsal Fin (C-C) ____________________ cm
Mass ____________________ gm (454 gm=1 lb; 28 gm=1 oz)
Fins: (number and description)
____ 1st Dorsal ____________________
____ 2nd Dorsal ____________________
____ Pectoral ____________________
____ Pelvic ____________________
____ Anal ____________________
____ Caudal ____________________
How many gill slits? ____________________
Are gill opercula present? ____________________
Where is the cloacal opening located? ____________________
Are claspers present? ____________________. If so, whre are they located? ____________________
Describe the surface color of specimen. ____________________
Describe the texture of the skin surface. ____________________

Open the mouth of the animal. Do you *see* any teeth? ____ With the mouth held open, feel for the presence of teeth with a finger-tip. Record findings:

Are teeth present on *both* the upper and lower jaws?

____

Draw a side view of specimen and label the following parts: nostril, mouth, eye, gill slits, all fins including the caudal (tail) fin, location of cloacal opening, claspers (if present), lateral line, spiracle. Significant external skin surface patterns should be shown. Indicate with lines "A-A", "B-B" and "C-C" where width measurements were taken.

Part III. Internal Observations

Expose the viscera by making a median longitudinal incision, ventrally, beginning at the cloacal opening and proceeding anteriorly. Describe the liver (size, number of lobes, texture and color). ________________

____

Remove the liver. How much does it weigh? ________

________________ gm What percentage of the total body mass (weight) consists of liver alone? ________ %

Is the alimentary canal longer than the body (exclude tail)? ________ . Locate and name the portions of the alimentary canal, beginning with the mouth:

Remove the entire digestive tract. Examine, then slit, the small intestine. Is a "scroll" present in this animal? ________

Locate the following organs. Indicate with a check (✓) if the organ(s) is (are) present in the specimen. Tell *where* they are located.

gall bladder ________ ________
pancreas ________ ________
spleen ________ ________
kidneys ________ ________
ovary (ovaries) ________ ________
testes ________ ________
rectal gland ________ ________

Locate the heart. Is there a partition separating the heart from the other organs?

length overall ________ cm
width overall ________ cm

Since the heart of embryo sharks is rather small, additional reading research will probably be necessary to complete the following questions.

Does the heart of the living shark contain both arterial and venous blood? ________________

Explain: ________________________

____

How many chambers are present? ________________

Name them: ________________________

Part IV. Brain Observation

Before dissecting, prepare a sketch of the shark (or related animal) from reading research. Label the following parts:

Olfactory lobe
Olfactory tract
Cerebral hemisphere
Optic lobe
Cerebellum
Medulla oblongata
Spinal cord
Cranial nerves

Remove as much skin and tissue as possible dorsally, between the eyes and over the general cranial area. Remember that the shark has a cartilaginous skeleton. Extreme care and patience is required. Expose as much of the brain as possible. It will be difficult to remove the brain, since it is held together very delicately.

With the help of the drawing, locate as many of the labeled parts as possible. Now, prepare another sketch of the brain of the specimen. How do the drawings compare?

How many cranial nerves are there?

Does the shark eye contain a crystalline lens?

Trace the optic nerve (Cranial Nerve No. II) to the brain. How many muscles move each eye?

QUESTIONS FOR CONSIDERATION

1. Trace the pathway of blood through the heart, gills and body of the shark.
2. What device(s) does the shark *lack*, making it necessary for the shark to be almost continually on the move?
3. Is the remora (*Echeneis naucrates*) a commentsal or a symbiont?
4. What is meant by (a) individual feeding pattern and (b) mob feeding pattern?
5. How do shark teeth differ between the species?
6. Can a shark be "trained"?
7. What purpose(s) does the "scroll" intestine serve?
8. Which species have a "spiral valve" intestine?
9. What purpose(s) does the lateral line serve?

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FIN RAY AND VERTEBRAE ANALYSIS - (Taxonomic Key - Bony Fish)

INTRODUCTION

Most of the available taxonomic keys to the fishes use such characteristics as the number of spines and rays in the dorsal and anal fins and the number of transverse rows of scales crossing the laterial line to separate species. It is difficult to find a color plate that illustrates the exact specimen at hand. This is especially true of the small brackish-water and fresh-water fishes of the Florida coast.

TO THE TEACHER

In fish identification there are relatively few current taxonomic keys available. Thus, quite frequently specimens are discarded because of no available means to establish identification. The method of staining the fish skeleton used here works well for small fish. By partially fleshing the skeleton, larger specimens can be stained successfully. The embryo of fish (see "Gambusia Embryology") can be placed in a plastic bag with fine holes punched in it and stained for fin and vertebrae development.

One must realize that a small or juvenile fish has a larger head and eye in proportion to the body than does the adult specimen. The number of fin rays and scale rows remains constant at all age levels.

It would be of great benefit to the student's understanding if preserved specimens of several months' standing were on hand to illustrate the color change brought about by preservatives when compared to live specimens in the aquarium.

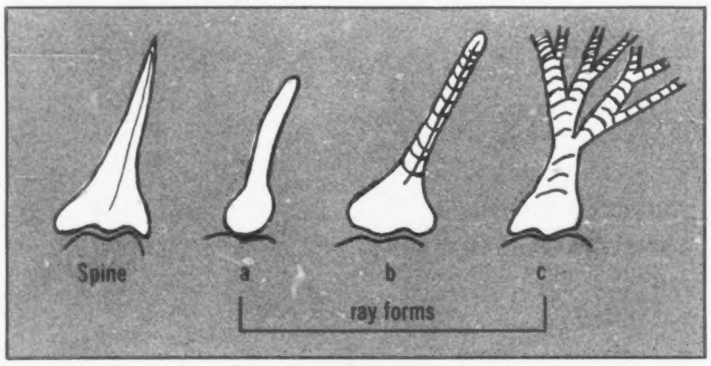
Seining in shallow brackish water should produce several different *Fundulus sp.* (killifishes) or *Gambusia sp.* Gobies, Blennies, and Pipefishes are good specimens, too

It is advised that wooden racks or trays be used for the containers. This will eliminate spillage of caustic material (KOH) and stain. It will also ensure that containers of solutions will not be shuffled out of sequence.

TO THE STUDENT

Using the color of fishes is not a reliable method for accurate identification because a large number of fish have the ability to modify their patterns. Secondly, others tend to fade and lose their color upon death and/or preservation. The dolphin with its brilliant blue-green and yellow flashing in the ocean becomes a dull grey on the dock. A large percentage of museum specimens tend to become bleached and colorless. The identification tag becomes the only clue remaining that the specimen was once a living animal.

This lab will give an adequate method of counting the number of rays present in the doral and anal fins of small fishes. These rays are frequently very difficult for the untrained eye to separate and count; this fact makes most available keys useless.



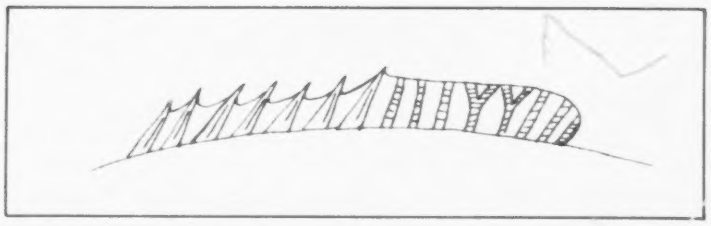
Spines and Soft Rays

The spines are smooth, pointed, solid, and not divided into segments. Soft rays are smooth, soft and not pointed, and may occur in three forms:

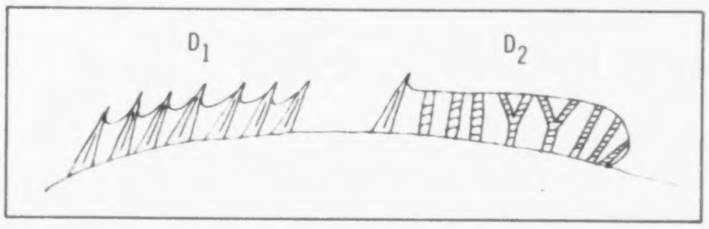
a) unbranched and non-segmented
b) unbranched and segmented
c) branched and segmented

[after Sterba (altered from Gunther) 1962]

A Roman numeral is used to represent the total spine number and an Arabic number to represent the total ray



number. In the illustration above, the code would be VII, 8. The numberal VII refers to seven spines; the number 8 refers to eight rays *(count the base of the rays)*.



Here the code would be VII - I.8. The VII represents the seven spines in D_1. The dash indicates that there are two fins. I,8 indicates that the second fin (D_2) has one spine and eight rays. Thus the fin is in two parts: One part has seven spines; the other part has one spine and eight rays.

The numbers are frequently preceded by a capital letter to indicate which fin is described (example: D = dorsal).

PURPOSE

To achieve an understanding of the taxonomic principles used in cataloging fishes and to analyze the skeletal structure of fishes.

MATERIALS

compound microscope
several species of fish
petri dish
white tray or a baby food jar
Alizarin Red S Stain
  Acetic acid – 5 ml.
  Glycerine – 10 ml.
  Choral Hydrate (1% solution) – 60 ml.
  Saturate above solution with Alizarin Red S Stain until no more stain will go into solution. Add 2 cc of this solution to 100 ml. of 4% KOH to produce the specimen staining solution.
Clearing Solution No. 1
  Glycerine – 20 ml.
  *4% KOH – 3 ml.
  Distilled water – 77 ml.
Clearing Solution No. 2
  Glycerine – 50 ml.
  4% KOH – 3 ml.
  Distilled water – 47 ml.
Clearing Solution No. 3
  Glycerine – 75 ml.
  Distilled water – 25 ml.
  4% KOH

PROCEDURE

1. Select two specimens each of 3 varieties of brackish water fish.

2. If the specimens are between two and four inches in length, gently remove the scales.

3. Place the fish in a white tray or enclose in a bottle with 4% KOH for bleaching. The KOH volume must cover the specimens used.

4. The specimens must be left two to three days in the sun or until completely bleached (translucent). Caution: If left too long, the specimen will tend to fall apart.

5. Immerse the fish in a container of the Alizarin Red S stain solution for approximately 24 hours or until the bones become a deep reddish-purple.

6. Place specimens in a beaker of fresh 4% KOH overnight to begin clearing process, then put fish in Clearing Solution No. 1 for two or three days.

7. Transfer specimens in Clearing Solution No. 2 for two or three days (longer if the tissue becomes more transparent).

8. Place fish in Clearing Solution No. 3 for two days.

9. Change specimens to pure glycerine for two days as the specimens are now reaching maximum transparency. In fact, they can now be stored in glycerine. Add a crystal of thymol to prevent fungus growth.

10. The fish are now placed in a petri dish half and observed under the compound microscope.

ORGANIZED DATA RECORDING

Record for each specimen:

  a. number of vertebrae
  b. number of fin rays in the dorsal fin
  c. number of fin rays in the anal fin

** dissolve 4 grams Potassium hydroxide in 20 ml. of distilled water, then add distilled water to make 100 ml. of solution.*

QUESTIONS FOR CONSIDERATION

1. Do all specimens of each type used possess the same number of anal and dorsal fin rays?

2. What physical functions do the fins have?

3. What differences occur within the skeletal structures that separate one species from another species in your sample?

4. What other anatomical and/or biochemical characteristics could be used to classify fishes?

LIMITATIONS AND SOURCES OF ERROR

a. Time requirement – the transfer of specimens requires 5 to 10 minutes each day, thus disrupting lecture time; specimens need to be close at hand to prevent lost motion.

b. Overstaining and failure to clear the specimen properly.

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DETERMINING THE AGE OF TELEOST FISH BY COUNTING SCALE RINGS

INTRODUCTION

In future years it may become very important to know the age composition of each type of fish. Through knowledge gained about the amount of growth attained each year and the life span of fishes, man can harvest the sea crop with greater efficiency.

Length-frequency distribution is the oldest age determination method in use. This involves utilizing large numbers of specimens and is based on the theory that all fish of one size are approximately the same age. This method has built-in disadvantages in that: (a) it does not separate older age groups, which tend to remain approximately the same size; (b) fish of the same size have a tendency to school together; (c) variable environmental conditions may allow one age group to grow at a faster rate or a reduced rate each year; (d) hatching does not always occur at the same time of the year; (e) it requires a large number of fish in a random sample.

The most positive and effective method of determining the age of fish is through the marking, release, and recovery of fish of a known age. The major disadvantages of this method are (a) cost of tagging; (b) time involvement; (c) the small number of specimens recovered and (d) damage from handling.

The generally accepted method of determining the age of fish is to count the annual layers deposited in the hard parts of a fish. The best hard structure to count is the scale, while the ear stone (otolith) and spine are in secondary positions. This method is dependent upon several items (a) accuracy is dependent upon interpretive ability and (b) the distinctiveness of the annual layers.

These layers in the hard parts of fishes are produced by metabolic processes and occur in a similar manner in the growth rings produced by trees each year.

TO THE TEACHER

In selecting specimens for use in this exercise, only fully-formed scales should be selected. It must also be realized that scale markings of fish in the tropical zone are sometimes imperfect because of the lack of a true winter season to slow the growth process.

Sometimes a false annulus is produced by females just prior to spawning as reabsorption occurs.

It may be to your advantage to select scales from large specimens for easier student observation – Snook, Bluefish, Jewfish, Sea Trout, Tarpon, etc.

Scales can be stored for use by placing in an evelope or pressed between pieces of paper. Mucus on the scale will hold the paper together. Species, place, weight, length, collector, sex, time and method of capture should be recorded with the scales.

The scales can be placed between two layers of thin, clear plastic and mounted in a 35 mm slide blank and projected upon a screen to produce the necessary magnification. This preparation lends itself to easy storing and cataloging.

Other methods of examination would include the overhead projector, opaque projector, and Glow Box (produced by I^2R Inc., Cheltenham, Pa.).

Some fish (catfish and eel) do not possess scales or the markings on the scales are not clear enough for interpretation. In these cases, the otolith is used. The otolith is a calcareous structure formed in the inner ear of the fish. Some thin otoliths can be read directly, others must be sectioned. In fact, some researchers also recommend grinding and polishing before interpretation.

TO THE STUDENT

In attempting to determine the age of fish by studying the structure of the scale, there are several definitions which must be cleared up before starting:

Annulus – The annual mark or zone found on the scales, vertebrae, otolith, or other hard portion of a fish, which is formed once a year. (Figure 2)

Ctenoid scale – The scale of a bony fish that possesses small sharp spines (ctenii), (Figure 1)

Cycloid scale – The scale of a bony fish without spines or ctenii. (Figure 1)

Ganoid scale – Thick plate-like scales. The annual rings are not well defined.

Focus – The small clear area near the center of a scale that represents the original scale of the young fish. (Figures 1 and 2)

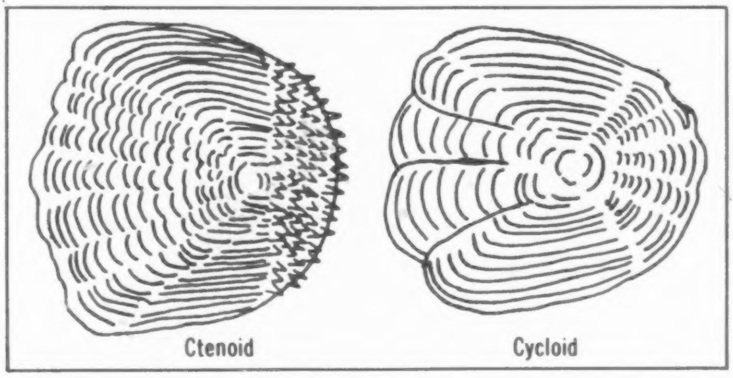

FIG. 1

The definitions given for cycloid and ctenoid are not fixed. There are many variations or degrees of spines or ctenii and the position of focus varies with each species of fish.

The annulus is recognized in one of the following ways: (a) "crossing over" where with the onset of fall or winter several ridges or circuli flare outward and end on the side of the scale rather than circle the focus; (b) "discontinuous circuli" – the individual circuli do not grow together in a complete line because the scale stops growing and (c) extreme crowding of the circuli which occurs just prior to a resumption of growth. (See Figure 2)

PURPOSE

To determine the age of a bony fish by examination of its scales.

MATERIALS

dissecting microscope or hand magnifier
slides
fish of several sizes or scales provided by the instructor
metric rule
projector

PROCEDURE

1. Remove a scale from each specimen to determine whether it is cycloid, ctenoid or ganoid.
2. If the scale is ctenoid, take scales for further examination from the region of the pectoral fin; cycloid scales should be removed from an area between the dorsal fin and the lateral line. Rounsefell and Everhart (1953).
3. Remove 3 scales from the indicated area from each specimen.
4. Mount the scales between two glass slides for observations under the microscope.
5. Record your observations.

ORGANIZED DATA RECORDING

1. Prepare a line drawing illustrating the general features of the scale: focus, annuli, circuli, ctenii (if present).
2. Using your metric ruler, determine the distance between the annuli on the scale (repeat for each scale of each specimen).
3. Each scale should be read *twice*, at different times, in order to arrive at an *accurate* interpretation of structure.
4. Counting one year of growth for each annulus, determine the age of each specimen.

QUESTIONS FOR CONSIDERATION

1. Is there a relationship between the type of scale (ctenoid or cycloid) and the soft or spiny state of the fin rays? If so, what?
2. How can you account for the varying distances between each annulus on the scales?
3. How much difference occurs in the distance between annuli on scales from the same specimen? Why does this occur, if present?
4. How is age determination of fishes useful in fisheries biology?

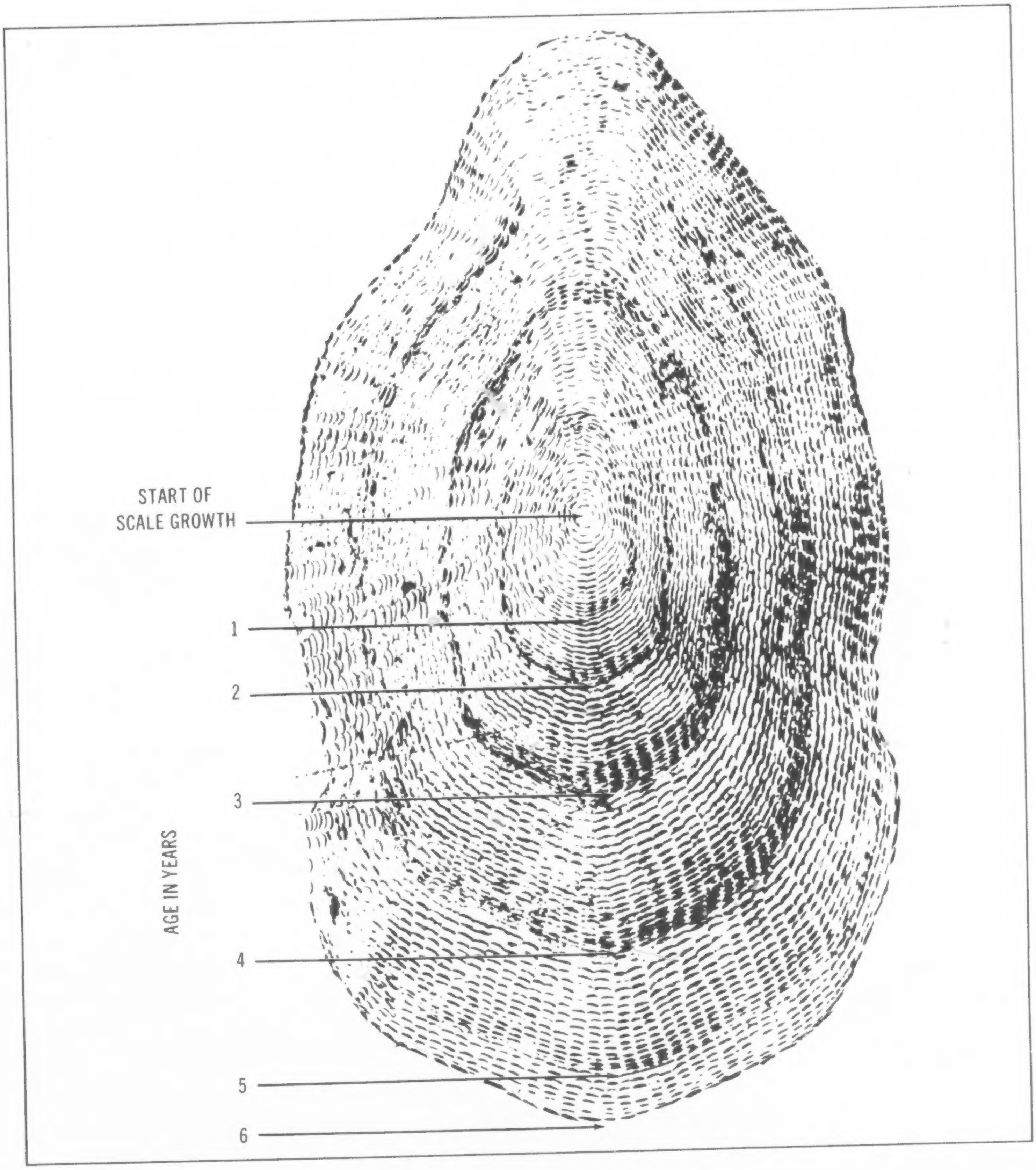
START OF
SCALE GROWTH
1
2
3
AGE IN YEARS
4
5
6

FIG. 2 Typical cycloid scale from a 6-year-old haddock. The more closely spaced rings (circuli) form darker year marks (annuli). The total annuli count determines the age of the fish. (U.S. Bureau of Commercial Fisheries)

LIMITATIONS AND SOURCES OF ERROR

1. Incorrect reading of scales.
2. Use of imperfect scales or scales that have been rejuvenated.
3. Some fishes show no definite annuli.
4. Errors in age determinations increase with the age of the fish.
5. Errors made in determining the location of the first annulus.

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THE FEEDING HABITS OF FISHES

TO THE TEACHER

This exercise involves one field trip (fishing trip) plus one to two periods in the laboratory. With proper consideration given to planning, this can be a fun lab as well as instructional. Due consideration should be given to the type of bait used in fishing, whether artificial, live, or frozen, in analyzing results.

If a fishing trip is not feasible, a seine haul along a shallow beach or estuary will provide the necessary specimens. If all else fails, go to a local fish house and secure fresh specimens.

This is a good time to collect scales, save small specimens for staining or build up the school museum.

TO THE STUDENT

Many species of fishes make use of a wide variety of food and some authorities state that fish will eat whatever is available. Lagler et al (1962) said, "As for the manner of feeding, only one broad common characteristic prevails – the food is taken into the mouth." However, each type of fish has some preference for food just as most people prefer steak to wieners. Some species of fish have adapted to such an extent that their mouth parts have changed to accept certain food or the stomach has evolved as a curved organ, or a grinding organ similar to the gizzard of a bird. *Hippocampus sp.*, *Syngnathoides sp.* and *Aeolisus sp.* are examples of a rather odd group which are stomachless. They swallow whole prey directly into the intestine. (Brown 1957).

The adaptations of the teeth to type of food consumed is apparent in many cases. The drum and sheepshead have large blunt crushing teeth for mollusks. Sharks, bluefish, and sea trout have sharp, inwardly-curving teeth for capture and retention of smaller fish. The herbivorous fish such as mullet have oral teeth, pharyngeal teeth, or a pyloric gizzard.

The exercise will attempt to correlate the type of teeth with the stomach contents or "who eats whom before who gets eaten". Occasionally a fish is caught with the tail of another fish protruding from its mouth. Careful attention should be paid to the possibility of finding fish tags on the specimens caught or in the stomachs analyzed.

PURPOSE

Examination of stomach contents and tooth shape to determine food selection preference.

MATERIALS

A. Field
- balance
- metric rule
- preservative*
- field notebook
- thermometer
- taxonomic keys
- syringe & needle
- fishing equipment & bait
- assorted bottles and jars
- labels
- knife
- watch
- sewing thread

B. Laboratory
- dissecting microscope
- compound microscope
- slides
- cover slips
- taxonomic keys
- dissecting instruments

PROCEDURE

A. In the field

1. On reaching the site chosen, each student is to record:

** 1 part 40% Formaldehyde, 7 parts seawater*

time of day
tide conditions
cloud cover
wind velocity
type of bottom
salinity (take a water sample)
temperature: air and water

2. Collect fish

3. As each fish is caught, record the following:

type of bait used
time caught
mass of fish
total length
mass of stomach
sketch of teeth

4. Slit the belly of the fish and remove the stomach.

5. Tie a piece of sewing thread around each end of the stomach.

6. Inject the stomach with preservative.

7. Either place the prepared stomach in an individual labeled jar or attach a label to the stomach and place in a group container.

8. Repeat step 1-7 for each fish caught.

B. Later back in the lab

1. Remove the stomach from its container and place it in a petri dish.

2. Slit the stomach and empty contents into petri dish.

3. Count and identify items present.

4. Remove a portion of the loose material trapped in the folds of the stomach wall, prepare a wet mount, and observe under the microscope.

6. Count and identify organisms.

GRAPHIC ANALYSIS

1. Graph the mass of a fish against its stomach mass. (hungry)

2. Calculate the frequency of occurrence for each organism identified. (number per animal)

3. Graph stage (high-mid-low) of tide to number of fish caught.

QUESTIONS FOR CONSIDERATION

1. What happened to the bait used to capture each fish?

2. Is or was there a correlation between time of day or tide and the fish caught? Explain.

3. What conclusions can be made concerning the feeding habits of species examined?

4. What organisms were most abundant and least abundant in the stomach of each observed species?

5. Describe any observed relationships between type of teeth and food consumed in the analyzed specimens.

6. A. What natural bait would be used to catch the following:

a. sheepshead
b. sea trout
c. snook
d. blue fish
e. mangrove snapper
f. pin fish
g. grouper
h. pompano
i. ______________________ (local specimens)

B. What artificial lure would be used for each of the above?

7. Why?

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DETERMINING SALINITY TOLERANCES OF LOCAL ORGANISMS

TO THE TEACHER

This laboratory exercise requires the student to design a simple experiment using previous experiences as a guide. He should have completed the laboratory, "Determination of Salinity of Sea Water", and be aware of the facets of a controlled experiment (control group, test group, variable, etc.).

Fiddler crabs, blue crabs, sea roaches, common minnows or killi-fishes, shrimp, snails or bivalves make excellent subjects.

It is suggested that students be briefed on the following points:

1. laboratory time available
2. space available
3. storage (gallon jars make excellent containers)

TO THE STUDENT

In any discussion regarding the forms of life found in fresh water and in marine water, one obvious generalization would probably arise – the flora and fauna inhabiting these two environments are different. Goldfish and water hyacinths have never, thus far, been discovered living in the ocean, nor have sailfish and sargassum weed been found in fresh water lakes or ponds. But someone would probably bring out the point that the above generalization does not pertain to certain cases. What of the dramatic spawning migrations of the fresh water eels of the genus *Anguilla,* who make their way to the depths of the ocean to lay their eggs and to die? What of the anadromous fishes (those fish which migrate from salt water to fresh water to spawn) such as the Pacific salmon and sea trout? And what of the tremendous numbers of organisms who inhabit coastal bays and lagoons, which are continually influenced by inflowing sea water and fresh water, droughts and heavy rainfall?

In some of these bays and lagoons, salinities have fluctuated from 0% to nearly 90%, within a relatively short period of time (Reid, 1955). What effect would this drastic change have on the local population? Would some organisms be expected to survive?

Marine biologists are devoting considerable time and effort to the question of salinity and its effects upon organisms. Some investigators found a definite correlation between salinity and gross size. It was also noted that the number of different species in estuarine areas declines along with a decrease in salinity Evidence has been accumulated to support the hypothesis that it is easier for marine animals to move into areas of lower salinity than for fresh water organisms to penetrate ocean waters.

Organisms which are able to tolerate a narrow salinity change are termed stenohaline, as compared to euryhaline forms which are able to thrive in water of greater salinity variation.

Studies have indicated that an organism's ability to tolerate varying salt concentrations are primarily due to the chemical nature of its body fluids, the efficiency and limitations of its osmoregulatory system under pituitory control, and the concentration of salts in the surrounding medium.

PURPOSE

To devise methods of establishing salinity tolerances of aquatic organisms.

PROCEDURE

1. Keeping in mind the limitations, if any, set forth by the teacher, design an experiment which would measure the adaptive ability of (*name of organism*) to salinity changes.
2. Include the following items in the plan:
   a. duration of experiment
   b. size of containers

c. number of specimens
d. types of daily observations to be made
e. types of graphs and/or tables to be constructed.

QUESTIONS FOR CONSIDERATION

1. How is an organism's internal environment affected by external changes in salinity?

2. How does the organism maintain its osmotic balance with the surrounding water? What structures are involved?

3. Could organisms increase their degree of tolerance to salinity changes if such changes occurred gradually rather than suddenly?

4. Why is it that sea gulls can drink sea water and survive . . . yet man would perish if he followed suit?

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LIGHT: THE IMPORTANCE OF THE STUDY OF THE PHYSICAL AND BIOLOGICAL PROPERTIES OF LIGHT IN OCEAN WATER

INTRODUCTION

The slogan for recent increased interest in the study of the oceans could well be "Man in the Sea". In the past man has operated from the surface of the sea. Now we are determined to *see* and work beneath the surface. We do this with cameras (still, movie, and TV), with our own eyes by getting man into various depths of the hostile undersea environment. The study of the behavior of light as it penetrates water can be divided into three general areas:

1. Descriptive Oceanography: this area is largely in the field of physics and is concerned with the behavior of light as it hits the air-water interface, and as it penetrates the water. This penetration of water by light is quite complex and involves reflection, refraction, diffraction, absorption, scattering and polarization. Add to this the constant changing of the intensity of the light due to changes in the position of the sun and variation in cloud cover and you can see how complex the study of light in water can be under natural conditions.

2. Biological Aspects: Photosynthesis: one factor affecting the population of micro-organisms in the oceans is the optical properties of water (plus naturally available food and water temperature). Many marine species, shrimp, etc., have diurnal migrations of considerable vertical distances. (The word *diurnal* means changes due to the position of sun.) The factors causing these migrations are still little known. A decrease in the surface temperature of the water near the surface at night might be a factor influencing vertical migration just as much as light. Not too many years ago the majority of marine biologists thought that most of the life in the oceans was near the surface where the light was most intense. As the result of a great deal of deep-sea research it is now known that planktonic organisms are found at all depths. Russell and Yonge (1936) and Nicol (1964) have excellent discussions of vertical distribution of marine animals.

3. The Effect of Light Penetration through Seawater as it Relates to Undersea Photography, TV, and Human Vision: the limiting factors of underwater visibility are due to two general factors – seawater is about a thousand times more dense than air and usually there is considerable organic and inorganic material suspended in the water. Recognition for distances of 100 feet underwater is possible only under ideal circumstances and sharp photographs are not possible beyond 30 feet. Special film, lens system, and lights have had to be developed. Dr. Harold Edgerton of the Massachusetts Institute of Technology was a pioneer in the development of deep sea photography.

TO THE TEACHER

The study of light and its influence on marine organisms shows how biology and other sciences are related. This is an example of the use of basic physics in a biological problem. The lab exercises suggested are open-ended. With simple basic apparatus, a wide variety of experiments may be conducted.

TO THE STUDENT

The experiment outlined gives ideas on how to build the apparatus. Use imagination to develop other ideas.

PURPOSE

What effect does light of varying intensities have on the vertical migration of marine animals? Most have read about some species of shrimp coming up near the surface at night. Do they do this because there is less light near the surface at night, or is the temperature lower at night, or is the food of the shrimp more abundant near the surface at night?

MATERIALS NEEDED

1. 100 ml. graduated cylinder
2. Enough black paper to cover the cylinder and to make a cover.
3. The *live* specimens to be used will depend on the local supply.
    a. If near the ocean, collect several pints of plankton. (See Lab. Exercise on Plankton – page 61).
    b. If not near the ocean, use the water flea (Daphnia) which can be collected in almost any clean pond.
    c. Brine shrimp *(Artemia)* can be reared in the lab.
    d. If a vertebrate specimen is needed, use guppies or any small fish which can be obtained in quantity.
    e. If a plankton net is not available, make one. Cut the top 10 or 12 inches from an old nylon stocking and sew it to a round piece of wire, tie a small plastic jar on the bottom of the net. Tie a string to the upper part of the net and it is ready.

PROCEDURE

1. Wrap the 1000 ml. graduated cylinder with the black paper and hold in place with rubber bands. At each 100 ml. mark cut out a flap in the paper which can be opened and shut. Make a light proof cover of the black paper for the cylinder.
2. Use a light source such as a microscope light; attach it to a ringstand so that it may be raised or lowered.
3. Fill the cylinder with salt or fresh water depending on the type of specimens being used.
4. Add organisms – use enough so you may easily see the action of your experiment. Make sure that organisms are evenly distributed throughout the water.
5. Put the cover on.
6. Turn on the light and open the flap at the 1000 ml. mark of cylinder. Let the light shine through for 10 minutes.
7. Open the top. Is there a concentration of animals in the lighted area? Try the same experiment at each level and make a record of observations. Vary the time that the light was at each opening. Is there any difference in the distribution of the animals?

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DETERMINATION OF POPULATION BY THE LINCOLN INDEX METHOD

INTRODUCTION

The study of populations of marine organisms is a "suit of many colors" because there is no single method which can be employed to fit all requirements. Animal abundance might be reported in terms of the number of individual organisms per unit area or volume. Perhaps an analysis of abundance in terms of mass (biomass) is desired. This is most extensively used in studies of fishes and plankton. Other requirements of study might require a population analysis of just certain-sized individuals or only those of a certain age.

The degree of accuracy in obtaining a population census, aside from depending upon method, is a matter of requirements. Area, weather, animal behavior, tides, currents, along with human energy and error produce countless variables which reduce the degree of accuracy in obtaining a population census.

Fully cognizant of the variables involved, assume that a certain ecological study requires that the population of a species of snail from Family Nassariidae be determined. Also assume that an adequate supply of animals is available. These snails are usually found along the water line in the bays and estuaries of Florida, rather than on the Gulf or Atlantic beaches. Random samples are desired, of course, but it is still difficult to be certain that this goal will be achieved.

> "The distribution of any species, plant or animal, is not random. It depends upon environment, although chance is in many cases the agent which brings it into the proper environment."
>
> (Benton & Werner, 1958)

TO THE TEACHER

By using snails referred to above, there is assurance of: (1) a relatively high degree of accuracy; (2) adequate numbers; and (3) ease in collecting, marking, and re-collecting. Using this lab as a beginning, it is possible to proceed into other animal and plant groups. For a quantitative size or mass determination refer to the section entitled "Statistical Methods".

For a class, the team approach (three-to-a-team) is advised. This exercise will require about an hour in the field and a few minutes at the lab for the precensus, and again for the census.

The precise species of snail depends upon the locality. In a pre-lab briefing students can be acquainted with the species selected.

Attention to the tides is mandatory. Try to be at the collecting site at lowest tide. Since there is an interval of three or four days between the precensus and the census, timing is important. It would be embarrassing to arrive on the site for the census at high water! Refer to the lab exercise on "Instruction in the Use of the Tide Tables".

If a series of counts is planned, a different color mark should be used each time.

For this lab, at least have the site already selected. The procedure will include the establishing of a base line. It may be wise to "map" the area. If so, sights and distances can be taken from this base line and used to complete a map of the area. A legend of the map could contain vegetation symbols, geological characteristics, scales, North marker, location and date.

The site selected should be representative of the population density of the general area.

TO THE STUDENT

Since populations of organisms vary with the season, and change from year to year, do not make the assumption that this census will remain constant.

Populations of these little snails sometimes number into the thousands, in a given area. To count each individual would be a hopeless task. The Lincoln Index (sometimes

Lindoln-Peterson) is the oldest and most widely-used indirect counting method. A known number of marked individuals will be released into the population. Later, a sample is collected.

> "Theoretically, the relationship between marked and unmarked individuals should be the same as in the population as a whole."
>
> (Benton & Werner, 1958)

The markings used must be permanent. Avoid putting the animal at a disadvantage when it is marked. Improperly marked, the animal may become vulnerable to predators. Paints, tags and clips often cause discomfort to the animal.

Establishing a base line will enable return to the original collecting site without marking trees or driving permanent stakes. The base line data will last as long as the reference points used.

PROBLEM

To estimate the population of *Nassaira sp.* along a given stretch of bay shoreline, by the Lincoln Index method.

MATERIALS

1 bottle red nail polish (each term)

1 bottle acetone, or polish-remover containing acetone (each team)

1 compass with bearing dial (one per class)
measuring tape or pre-measured string (for the class)

collecting bags or buckets (each student)

3 or 4 Q-tips (each team)

PROCEDURE

This is a *team* effort. Individual responsibility to the class dictates the best individual effort. This is to be total count census of a species of snail (Family Nassariidae).

PART I – *The Precensus*

1. Establish a base line, 10 to 20 meters long, at right angles to the shore-line. Select a durable-looking tree and sight the base line from this point. If a natural, fixed point is not available, it will, of course, be necessary to drive a stake or build some sort of marker. Drive temporary stakes to mark the base line. Record the bearing of base line and measure its length.
2. Measure 30 meters distance each way from the base line and drive temporary stakes.
3. All members of the class will station themselves, more or less evenly, throughout the area.
4. Each student will then pick up approximately 50 live snails (any size).
5. Teams will then group and mark these snails. So that the nail polish adheres properly, clean a section of the shell with a Q-tip and acetone. Let the acetone dry, then apply the red polish. Avoid the operculum; apply the acetone and polish to the pointed end of the snail.
6. Each team will record the number of *marked live* snails.
7. Members of each team will disperse throughout the area and scatter (do not throw) the snails back into the population.
8. Pull up the temporary stakes.
9. Return to the area after three days.

PART II –

PART II – *The Census*

1. Upon returning to the precensus site, re-establish the base line and distances, as before.
2. All students will disperse again through the collecting site and gather snails. Pick up living snails, at random, and try to be objective in your selection. In other words, do not concentrate on just those that are marked! When each student has picked up approximately 50 snails, regroup into teams.
3. Each team is to record:
   (a) the number of marked snails
   (b) the number of unmarked snails
4. Recover the stakes and release the animals.

INSTRUCTIONS FOR DATA ANALYSIS

1. Recall the following:
   a. Number caught and marked in the precensus = ______
   b. Number caught in the census which were marked = ______
   c. Number caught in the census which had no mark = ______

2. Apply these data to the Lincoln Index formula:

$$\frac{\text{Caught and marked: precensus}}{\text{Others present but not caught during precensus}} = \frac{\text{Marked and caught: census}}{\text{Unmarked and caught: census}}$$

Solve by proportion to find "others present, but not caught, during precensus". ans. ____________

---

3. To arrive at a determination of the estimated total population, add:

____________ = Caught and marked, precensus

+ ____________ = *Others present, but not caught during precensus*

ans: ____________ = Estimated total population

QUESTIONS FOR CONSIDERATION

1. Can you think of some variations on the lab using the same animal?

2. Name other animals whose population counts could be determined by the Lincoln Index. What limitations would be imposed for each animal you select?

3. What vocations might utilize the Lincoln Index method?

4. What does larithmics mean?

5. What is meant by eugenics; euthenics?

6. What is meant by natality?

7. Make a list of possible sources of error in this lab. For each source of error listed, prescribe a possible remedy.

8. Are there any ways other than the Lincoln Index to determine total populations? List the advantages of alternate methods.

9. Relate tagging, conservation and the Lincoln Index method!

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ANALYSIS OF MARINE POPULATIONS

TO THE TEACHER

This exercise lends itself best to a combination field-laboratory study. Samples may be taken during one field trip or even at regular intervals for several months (to determine if seasonality influences the composition of seaweed populations), preserved in formalin or alcohol and analyzed at a convenient time in the laboratory. Seaweed clumps are not common to all coastal areas throughout the year. Become familiar with the seaweed influx (primarily *Sargassum*) at local collecting sites.

If *Sargassum* clumps are not available when needed, other potentially interesting niches can easily be substituted (floating or submerged wood, sponges, rocks, tidal pools, etc.). Emphasis should be placed on sampling and collecting techniques, limitations, and methods of analyzing data.

TO THE STUDENT

Of the many diverse niches in the marine environment probably the most bizarre are floating *Sargassum* clumps. *Sargassum*, or gulfweed, is the commonest type of brown algae in Florida waters and can be easily recognized by the conspicuous berry-like floats scattered throughout its foliage. Tremendous quantities of gulfweed are found in the Atlantic Ocean and in the Gulf of Mexico.

The *Sargassum* jungle teems with life of all sorts. Even the small island-like clumps which drift about close to shore carry with them an amazing assortment of living creatures. Minute shrimp, barnacles, crabs, slugs, snails, bryozoans, worms and even fish comprise the majority of creatures which inhabit the entangled masses of stems and leaves. Many of these animals have become specially adapted to their seaweed home. Because of the unique mode of life of the *Sargassum* population, that of being restricted, for the most part, to their floating home on the surface of the ocean, their distribution patterns are dependent upon the direction of surface currents and prevailing winds. Salinity, temperature, and the abundance of food also affect the distribution and faunal composition of the niche.

PURPOSE

The purpose of this field-laboratory study is to analyze the faunal population of *Sargassum* clumps.

MATERIALS LIST

Field

plastic specimen bags or glass jars
preservative
labels
wide diameter scoop net with fine mesh net

Laboratory

dissecting microscopes or
magnifiers
pans
forceps
sorting jars
rulers and balances

PROCEDURES

. . . In the field:

1. Carefully scoop up each *Sargassum* clump and deposit its entire contents into collection container. Rinse out net into container to insure complete sample.
2. Add preservative, close and tag. Indicate on tag or notebook the following data: date, time, location, prevailing wind, tide, wave activity, air temperature, water temperature, and salinity (if questionable).
3. Secure as many clumps as needed. (One clump per student or team.)

. . . In the laboratory:

1. Empty sample into a pan, rinsing out container to include possible clinging specimens.
2. Examining every part of *Sargassum*, remove all attached and free organisms.

3. Record the following data for each clump:
    a. Total number of organisms
    b. Numerical breakdown of population according to taxonomic groups
    c. Total wet weight of Sargassum clump
    d. Total wet weight of population
    e. Weight and length breakdown of taxonomic groups
    f. Indicate whether attached or free
    g. Indicate specialized adaptations of taxonomic groups

SUGGESTED MATHEMATICAL COMPUTATIONS

1. Compute ratio of clump weight to population weight.
2. Compute per cent of total population for each taxonomic group by numbers and weights.
3. Frequency distribution of taxonomic groups in *Sargassum* clumps.
4. Weight-length ratios of taxonomic groups in each clump.
5. Theoretical food-chain relationship of *Sargassum* animals.

QUESTIONS FOR CONSIDERATION

1. Which adaptations did members of the *Sargassum* population exhibit that would increase their chances of survival?
2. What is your opinion of the methods used in collecting *Sargassum* clumps?
3. What types of predators would *Sargassum* animals have to cope with?
4. How are these creatures affected by ocean waves and storms?

SUGGESTED STATISTICAL COMPUTATIONS

1. Determine mean and standard deviation values for:
    a. Total populations of clumps (number and weights)
    b. Taxonomic groups (number and weights)

LIMITATIONS AND SOURCES OF ERROR

1. Size of the sample (number of clumps)
2. Accuracy of sampling technique
3. Geographical size of the collecting area
4. Time of day
5. Seasonal limitations
6. Human error

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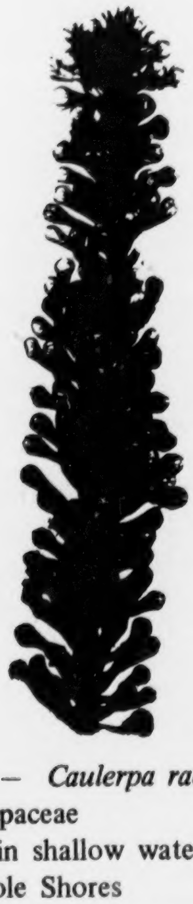
PREPARATION OF HERBARIUM MOUNTS

TO THE TEACHER

In collecting algae in the field, all specimens from a particular habitat should be placed in a plastic bag and numbered to match a data number from the field notebook.

This exercise should begin with mounting and pressing at the end of the week and drying over the week-end in dry chamber or oven. (see page 149). Cards can be dried in an incubator for the week-end. Classification can begin on Monday and continue for as long as necessary or desired. Mounts can be prepared individually or as a composite.



Technical Name – *Caulerpa racemosa* V. *laetervirns*
Family – Caulerpaceae
Habitat – reefs in shallow waters
Locale – Seminole Shores
Description – dense, robust, erect
Date – May, 1969
Collector – Jacque Penick

This lab may be run concurrently with the analysis of floating seaweed populations if desired.

These techniques are applicable to fresh water algae and assorted water plants as well as marine algae.

It is suggested that the teacher choose the best specimens each year and use these to build up a school phycology collection.

The teacher may desire to use standard herbarium paper or some other cardboard stock rather than file cards in order to secure larger specimen mounts.

TO THE STUDENT

As a coming source of food for mankind, algae cannot be overlooked. At the present time seaweeds, giant kelp, are being collected and converted to flour. The Japanese are raising *Chlorella* to be used as a protein, fat, and vitamin supplement for their diets.

Apart from its use for the culture of micro-organisms, the extracted agar from seaweeds has a variety of uses. Among these can be included the canning of fish, the sizing of fabric, in paper and glue manufacture, to add gloss and stiffness to leather, in cosmetics and medicines, and as a thickening agent in ice-creams, sherbets, and pastries.

PURPOSE

To prepare herbarium mounts of marine algae.

MATERIALS LIST

plant press
plastic bags and ties
tags (white paper)

shallow pans or trays
probes
scissors
5 x 8 file cards or herbarium sheets
wax paper
cellophane wrap
newspaper
drying box
assorted algae

PROCEDURE

1. Remove a specimen from a numbered bag and record this number on 5x8 unlined file card.

2. To a shallow, flat pan add water to a depth of 1/4 inch.

3. Place the algae in the pan and tease it into flat position.

4. Select a representative portion to use as a mount.

5. Slip the card under the specimen and arrange the specimen into desired position.

6. Remove the mount by grasping a corner and raising gently to drain the water. A second method is to mount the paper on a floating board and place in water for mounting.

7. Place the mounted algae into a plant press using alternate layers of newspapers, the algae mount, wax paper, newspaper.

8. Place the press in a drying box or a hot dry place.

9. Change the newspaper and wax paper every 6-12 hours or as often as possible to at least once a day.

10. When the mount is dry, cover it with a sheet of wrap slightly larger than the mount and overlap the self-adhering portions on the back of the card.

11. Mount the preparation on a sheet of 8-1/2 x 11 stock and record collecting data from corresponding number records. (see form this page)

12. Secure dichotomous keys and/or refer to the school collection for identification. Record on the form below.

Organized data recording:

Technical name –

Common name –

Family –

Habitat –

Locale –

Description –

Date –

Collector –

QUESTIONS FOR CONSIDERATION

1. What is the educational value of a herbarium collection?

2. How are or can algae be used as indicators for the animal populations or mineral deposits?

3. Of what value is the information recorded on the prepared algae mount?

4. What future benefits to man may be derived from the culture of algae?

LIMITATIONS AND SOURCES OF ERROR

1. Algae need more pressure applied than standard botanical mounts.

2. If the newspaper and wax paper are not changed on a regular basis the algae will begin to adhere to the wax paper.

3. Extreme care should be used in selecting the portion of algae for mounting.

4. Wasted time if not properly scheduled.

RESOURCE MATERIALS

Algae will be found almost universally distributed through the shallow, off-shore areas, on reefs, around mangrove hammocks, bays, and estuaries.

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Smith, Gilbert M., 1951, *Manual of Phycology*, Chronica Botanica Company: Waltham, Mass.

Taylor, W. R., 1960, *Marine Algae of the Eastern Tropical and Subtropical Coasts of the Americas*, University of Michigan Press: Ann Arbor, Michigan.

Verville, Ad. D. and J. Feldman (Eds.), 1964, *Seaweed Symposium*, Macmillan: New York.

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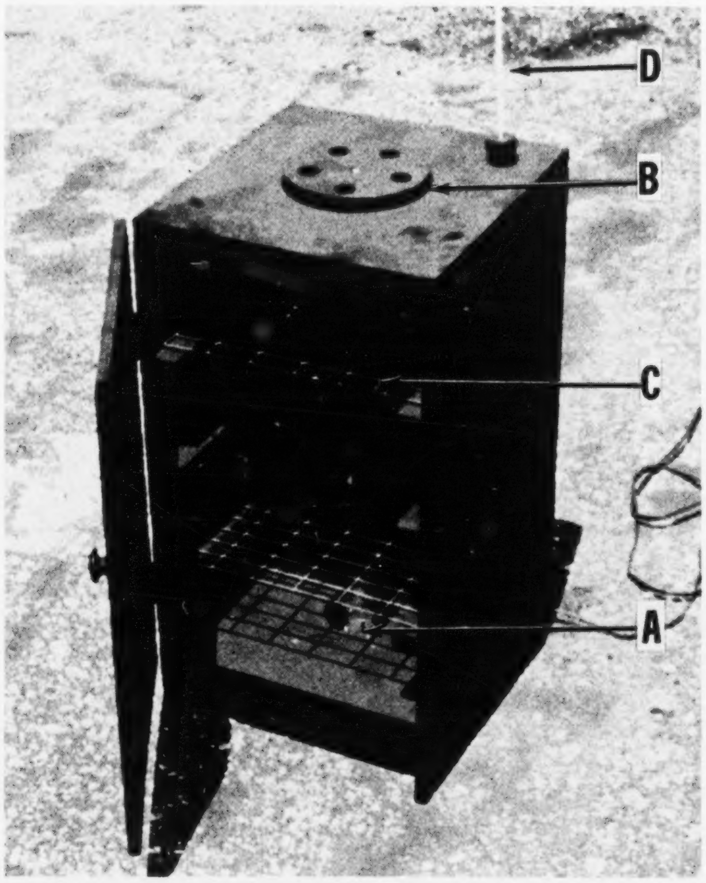
HERBARIUM AND INSTRUMENT DRYING BOX



A rectangular box of 1/2" plywood built in size to accommodate one or more herbarium presses. The size is dependent upon available space and need.

Item A in the illustration represents a 40 to 75 watt light bulb which provides heat for drying.

Item B is a series of air holes for moisture-laden air to escape. These should be drilled to some available stopper size as a No. 5, 6, or 7, or wheel-shaped closure as illustrated. Item C represents a wire shelf. This should be placed a little below the center of the box, so that items placed on it will be in the approximate center. If desired, more shelves can be added. Discarded refrigerator racks can be used as shelves. Item D represents a thermometer.

The box illustrated here is 28" w x 28" DX 36" H. The closure wheel is 10" in diameter.

The drying box is also effective for drying instruments. If the instruments are rinsed in running water after use with salty marine specimens, much of the rusting usually associated with these operations can be avoided.

TABLE OF CONVERSION FACTORS*

(U.S. units to other U.S. units)

Length

1 inch (in.) = 0.083 ft.
1 foot (ft.) = 12 in.
1 yard (yd.) = 36 in.
= 3 ft.
1 fathom = 6 feet
1 statute mile = 5,280 feet
1 nautical mile = 6,080 ft.
= 2,026.7 yd.

Area

1 sq. in. = 0.0069 sq. ft.
1 sq. ft. = 144 sq. in.
1 sq. yd. = 1,296 sq. in.
= 9 sq. ft.
1 acre = 43,560 sq. ft.
= 0.00156 sq. mi.
1 sq. mile = 640 acres

Volume (cubic measurements)

1 cu. in. = 0.00058 cu. ft.
1 cu. ft. = 1,728 cu. in.
= 29.92 quarts
= 7.48 gallons
1 cu. yd. = 27 cu. ft.

Capacity (liquid measure)

1 pint (pt.) = 16 fluid ounces
= 28.88 cu. in.
1 quart (qt.) = 2 pt.
= 57.75 cu. in.
1 gallon (gal.) = 4 qt.
= 231 cu. in.

Weight (avoirdupois)

1 ounce (oz.) = 0.0625 lb.
1 pound (lb.) = 16 oz.
1 short ton = 2,000 lb.

Weights of water

1 quart = 2 pounds (fresh water)
1 cu. ft. = 62.4 lbs. (fresh water)
= 64 lbs. (sea water)

Pressure

1 pound per square inch (p.s.i.) = 2.31 feet of fresh water
= 2.25 feet of sea water
= 0.068 atm.
= 2.036 in. Hg.
1 atmosphere (atm.) = 14.696 p.s.i.
= 29.92 in. Hg.
= 33.9 ft. of fresh water
= 33 ft. of sea water
1 foot of sea water = 0.445 p.s.i.
1 inch of mercury (in. Hg.) = 0.491 p.s.i.
= 1.133 feet of fresh water
= 13.60 inches of fresh water

Data from: U.S. Government Printing Office. 1963. U.S. Navy Diving Manual. Part I. Government Printing Office: Washington, D.C.

TABLE OF CONVERSION FACTORS*

(Metric units to other metric units)

Length

1 millimeter (mm.) = 0.1 cm.
= 0.001 m.
1 centimeter (cm.) = 10 mm.
= 0.01 m.
1 decimeter (dm.) = 100 mm.
= 10 cm.
= 0.1 m.
1 meter (m.) = 1000 mm.
= 100 cm.
= 10 dm.
= 0.001 km.
1 kilometer (km.) = 1000 m.

Volume and Capacity

1 cubic centimeter (cc.)
(or 1 millimeter (ml.)) = 0.001 liter
1 liter (l.) = 1000.027 cc.
= 1000 ml.
= 0.001 cu. m. (m.^{3})
1 cubic meter (m.^{3}) = 1000 l.

Area

1 sq. cm. (cm.^{2}) = 100 mm.^{2}
1 sq. m. (m.^{2}) = 10,000 cm.^{2}
1 sq. km. (km^2) = 1,000,000 m.^{2}

NOTE—European usage employs a comma where we use a decimal point and a period where we use a comma (in large numbers).

Weight

1 milligram (mgm.) = 0.001 gm.
1 gram (gm.) = 1000 mgm.
= 0.001 kg.
1 kilogram (kg.) = 1000 gm.

Weights of fresh water

1 cc. or 1 ml. = 1 gm.
1 liter = 1 kilogram

General System of Multiples

Multiple	Prefix	Symbol
10^{12}	tera	T
10^{9}	giga	G
10^{6}	mega	M
10^{3}	kilo	k
10^{2}	hecto	h
10	deka	da
10^{-1}	deci	d
10^{-2}	centi	c
10^{-3}	milli	m
10^{-6}	micro	μ
10^{-9}	nano	n
10^{-12}	pico	p
10^{-15}	femto	f
10^{-18}	atto	a

Pressure

1 gram per square centimeter (gm./cm.^{2}) = 0.001 kg./cm.^{2}
= 1 cm. of fresh water
1 kilogram per square centimeter (kg./cm.^{2}) = 1000 gm./cm.^{2}
= 10 meters of fresh water
= 9.75 meters of sea water
= 73.56 cm. Hg.
= 0.968 atm.
1 centimeter of mercury (cm. Hg.) = 13.6 gm./cm.^{2}
= 13.6 cm. of fresh water
1 centimeter of fresh water = 1 gm./cm.^{2}
1 atmosphere = 1.033 kg./cm.^{2}
= 760 mm. Hg.

*Data from: U.S. Government Printing Office. 1963. U.S. Navy Diving Manual. Part I. Government Printing Office: Washington, D.C.

TABLE OF CONVERSION FACTORS*

(U.S. units to metric units)

Length

1 inch = 25.4 mm.
= 2.54 cm.
1 foot = 30.48 cm.
= 0.3048 m.
1 statute mile = 1.609 km.
1 nautical mile = 1.853 km.

Area

1 sq. in. = 6.45 cm.^{2}
1 sq. ft. = 929.03 cm.^{2}
= 0.0929 m.^{2}

Volume and capacity

1 cubic inch = 16.39 cc.
1 cubic foot = 28,317 cc.
= 28.317 liters
= 0.028317 cu. m.
1 quart = 0.946 liter

Weight

1 ounce = 28.35 gm.
1 pound = 453.6 gm.
= 0.454 kg.
1 short ton = 907.2 kg.

Pressure

1 p.s.i. = 70.3 gm./cm.^{2}
= 0.0703 kg./cm.^{2}
= 0.703 meter of fresh water
= 5.17 cm. Hg
1 in. of fresh water = 25.4 mm. water
= 2.54 gm./cm.^{2}
1 in. of mercury = 25.4 mm. Hg
= 34.54 gm./cm.^{2}

TABLE OF CONVERSION FACTORS*

(Metric units to U.S. units)

Length

1 cm = 0.394 in.
1 meter = 39.37 in.
= 3.28 ft.
1 kilometer = 0.621 mi.

Area

1 cm.^{2} = 0.155 sq. in.
1 m.^{2} = 10.76 sq. ft.
1 sq. km = 0.386 sq. mi.

Volume and capacity

1 cc. or ml. = 0.061 cu. in.
1 cu. m. = 35.31 cu. ft.
1 liter = 61.02 cu. in.
= 0.035 cu. ft.
= 33.81 fl. oz.
= 1.057 quarts

Weight

1 gram = 0.035 oz.
1 kg = 35.27 oz.
= 2.205 lb.

Pressure

1 gm./cm.^{2} = 0.394 inch of fresh water
1 kg./cm.^{2} = 14.22 p.s.i.
= 32.8 feet of fresh water
= 28.96 inches of mercury
1 cm. Hg = 0.193 p.s.i.
= 0.446 foot of fresh water
= 0.394 inch of mercury
1 cm. of fresh water = 0.394 inch of fresh water

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*Data from: U.S. Government Printing Office. 1963. U.S. Navy Diving Manual. Part I. Government Printing Office. Washington, D.C.

PERIODICALS, NEWSLETTERS, JOURNALS

The periodicals and newsletters listed below:

(a) are devoted to marine science research

(b) are industrial or sales organization, inhouse journal

(c) contain popular non-technical articles on marine science

(d) have occasional articles, technical or popular, in marine science

(e) contain news of what is currently happening in research and industry

These letter designations are placed beside each title to identify its possible use by the marine science teacher and student.

The items listed as free are free only to qualified scientists, teachers, and researchers. Students are *not* invited to apply for them, with the possible exception of "Sea Secrets".

The teacher may find he does not qualify to receive free copies in all cases and should be prepared to pay subscription costs upon request.

It may be advisable to write for a sample copy of a publication to determine whether or not it fits the needs of the local situation, prior to paying a subscription fee.

The American Biology Teacher (d)
National Assoc. of Biology Teachers
NABT Executive Offices
1420 N. Street, N.W.
Washington, D.C. 20005
$8.00 9/year

Audubon (d)
National Audubon Society
Membership Dept.
1130 Fifth Ave.
New York, N.Y. 10028
individual–$7.00
institution–$5.00 9/year

Biological Bulletin (a & d)
The Marine Biological Laboratory
Woods Hole, Mass.
$18.00 4/year

Bulletin of Marine Science (a)
University of Miami Institute of Marine Science
1, Rickenbacker Causeway
Virginia Key
Miami, Fla. 33149
$12.50 4/year

Caribbean Journal of Science (d)
Institute of Caribbean Science
University of Puerto Rico
Mayagues, Puerto Rico
articles in English and Spanish
$2.00 4/year

Carolina Tips (b & d)
Carolina Biological Supply Co.
Burlington, N.C. 27215
free 10/year

Copeia (a & d)
American Society of Ichthyologists and Herpetologists
Division of Reptiles and Amphibians
U.S. National Museum
Washington, D.C. 20560
$10.00 4/year

Davy Jones Newsletter (b)
Edgerton, Germehausen, & Grier Inc.
160 Brookline Ave.
Boston 15, Mass.
free irregular

Deep Sea Research (a)
Pergamon Press
122 East 55th Street
New York 22, N.Y.
$10.00 6/year

Ecology (d)
Ecological Society of America
Duke University Press
Durham, N.C. 27706
$12.00 6/year

Ecological Monographs (d)
Duke University Press
Durham, N.C. 27706
$6.00 4/year

Environmental Science & Technology (a & d)
American Chemical Society
1155 Sixteenth St. N. W.
Washington, D.C. 20036
$7.00 12/year

Florida Conservation News (c)
Florida State Board of Conservation
Tallahassee, Florida
free 12/year

Florida Health Notes (d)
Florida State Board of Health
Publication Office
Box 210
Jacksonville, Fla. 32201
free 12/year

The Florida Naturalist (d)
Florida Audubon Society
Cor. South Lack Sybelia Drive & East Street
Maitland, Fla. 32751
$5.00 12/year

Florida Wildlife (d)
Florida Game and Fresh Water Fish Commission
Tallahassee, Fla. 32304
$2.50 12/year

Frontiers (d)
The Academy of Natural Sciences of Philadelphia
19th and The Parkway
Philadelphia, Pa. 19103
$2.50 5/year

International Marine Science (a & e)
UNESCO Publications Center
317 East 34 Street
New York, N.Y. 10016
$2.50 4/year

Journal of Limnology and Oceanography (a)
Allen Press
Lawrence, Kansas
$10.00 6/year

Journal of Marine Research (a)
Bingham Oceanographic Laboratory
Yale University
New Haven, Conn.
$10.00 3/year

Journal of Marine Science (a)
Librarian
Institute of Marine Science
Port Aransas, Texas 78373
$4.35 1/year

MTS Memo (e)
Marine Technology Society
1730 M Street N.W.
Washington, D.C. 20036
$15.00 12/year & Special Issues

Marine Science Newsletter
Orange County Schools
Santa Anna, California
Free 6/year

Maritimes (c)
University of Rhode Island
Graduate School of Oceanography
Wakefield, Rhode Island
free, limited distribution, 4/year

NASCO Notes (e)
Committee on Oceanography
National Academy of Sciences
Washington, D.C. 20418
free, limited distribution, irregular

National Fisherman
22 Main Street
Camden, Maine 04843
$4.00 13/year

National Geographic (d)
National Geographic Society
17th and M Streets NW
Washington, D.C. 20036
$6.50 12/year

National Wildlife (d)
National Wildlife Federation
Membership Services
1255 Portland Pl.
Boulder, Colo. 80301
$5.00 6/year

Natural History (c & d)
The American Museum of Natural History
Central Park West at 79th Street
New York, N.Y. 10024
$7.00 10/year

Naval Oceanographic Newsletter (e)
U.S. Naval Oceanographic Office
Washington, D.C.
free, limited distribution, irregular

News
National Oceanography Association
1900 L Street NW
Washington, D.C. 20336
$10.00 12/year

Newsletter (e)
National Oceanographic Data Center
Washington, D.C. 20390
free 10/year

Ocean Industry (e)
Gulf Publishing Co.
Box 2608
Houston, Texas 77001
Free/$12.00 12/year

Oceanology International (a & e)
Oceanology, Inc.
Industrial Research Bldg.
Beverly Shores, Ind. 46301
free to qualified scientists
$10.00 12/year

Oceans Magazine
P. O. Box 121
Des Moines, Iowa 50301
$12.00 12/year

Ocean Systems News (b)
Ocean Systems, Inc.
270 Park Ave.
New York, N.Y. 10017
free–limited distribution

Offshore (a & b)
International Trade Publications
Conroe, Texas
concerned with petroleum
price unknown 12/year

The Plaster Jacket (d)
Florida State Museum
University of Florida
Gainesville, Fla. 32601
free, irregular

The Progressive Fish-Culturist (d)
U.S. Dept. of the Interior
Fish and Wildlife Service
Bureau of Sport Fisheries and Wildlife
Washington, D.C. 20402
$1.00 4/year

Science (d)
AAAS
1515 Massachusetts Ave. NW
Washington, D.C. 20005
$8.50 weekly

The Scince Teacher (d)
National Science Teachers Assoc.
1201 Sixteenth St. NW
Washington, D.D. 20036
$8.00 9/year

Scientific American (d)
Scientific American, Inc.
415 Madison Ave.
New York, N.Y. 10017
$7.00 12/year

Sea Frontiers (c)
International Oceanographic Foundation
Institute of Marine Science
1 Rickenbacker Causeway
Miami, Fla. 33149
$6.00 6/year

The Seahorse (b)
Oceanographic Engineering Corp.
Hydro Products Division
P. O. Box 10766
San Diego 10, Calif.
free 4/year

Sea Secrets (c)
International Oceanographic Foundation
1 Rickenbacker Causeway
Virginia Key
Miami, Fla 33149
free 12/year

Shore and Beach (a & c)
American Shore and Beach Preservation Assoc.
Washington, D.C.
price unknown 12/year

Skin Diver (c)
Skin Diver Publications
Lynwood, Calif.
$7.00 12/year

Turtox News (d & b)
General Biological Supply House
8200 South Hoyne Ave.
Chicago, Ill. 60620

Undersea Technology (a & b)
Compass Publications
617 Lynn Building
111 No. 19th Street
Arlington, Va. 22209

free to qualified oceanographers
$8.00 12/year

Under Water Naturalist (c)
American Littoral Society
Sandy Hook Marine Laboratory
Highlands, New Jersey
$5.00 to members, 4/year

The Underwater Reporter (c & e)
Underwater Society of America
427 Chestnut St.
Philadelphia, Pa. 19106
$6.50 with associate membership 4/year

FILM LIST

This revised list includes a special inclusion of **FREE** films. These are from governmental and service agencies.

An Acid-Base Titration 30 min., sd, color, 16mm, E.B.F. Burets and the techniques for filling and operating are shown. A titration is carried out.

Adaption to Marine Environment color. McGraw-Hill Text-Films. 19 min. Experiments showing how a physiologist determined the mechanisms enabling a frog from Thailand to withstand drastic changes from living in and out of water and also in fresh or brackish water.

Alvin Sr. (Flying at the bottom of the sea.) Filmtronics Lab., Inc. 30 min., sound, color. Description of construction and operation of the deep sea research submarine, Alvin.

Atmosphere and Its Circulation 11 min., sd, b&w, 16mm, E.B.F., 1945. Explains characteristics of the atmosphere, and the factors which cause the circulation of the air and wind.

The Artic Region and Its Polar Bears 28 min., sd, color, 16mm, Disney, 1964. A photographic study of the polar bear and his home in the Arctic wilderness. Shows families of walrus as the thaw reopens the channels.

Audubon and the Birds of America 16 min., sd, color, 16mm, Coronet, 1957. Life of Audubon: his struggles to compensate for repeated business failure by taking intense interest in painting wildlife.

Beach and Sea Animals 12 min., sd, color, 16mm, E.B.F., Revised Edition. Uses underwater close-up photography to examine the habits and characteristics of invertebrate animals dwelling on or near the beach.

The Beach: a River of Sand 20 min., sd, color, 16mm, E.B.F., 1966. Describes the flow of a beach, and explains methods of measuring the movement of the sand.

Biochemistry – A General View 30 min., sd, b&w, 16mm, E.B.F. Biochemistry is the chemistry of living things. Fifteen or more elements have been found to be essential in animals and plants.

The Bird Community 12 min., sd, color, 16mm, Moody Inst., 1955. Presents a study of the community life of the birds inhabiting the Midway Islands in the central Pacific.

Birds of the Marshes 11 min., sd, color, 16mm, Coronet, 1946. Reveals the usually secluded lives of various marsh birds. Shows bittern, wrens, terns, rails and grebes.

Birds of the Inland Waterways 11 min., sd, color, 16mm, Coronet, 1946. Presents various birds of inland waterways and their native habitats. Shows sandpiper, ibis, avocet, etc.

Birds That Eat Fish International Film Bureau. 6 min., sd, color. Physical structure enables some birds to eat fish. Includes heron, Kingfisher, esprey, cormorant, grebe, loon.

Bird Wing Adaptions International Film Bureau. 17 min., sd, color. Shows how each bird developed its wings to help it survive in its habitat; good for teaching evolutionary adaptation.

The Birth of a Florida Key 14 min., sd, color, 16mm, DPM Prod., 1953. Nature study describing origin of the Florida Keys from seed pods of mangrove tree to fullsized Keys.

Blessing from the Sea FREE
Florida Phosphate Council
P.O.Box 1565
Lakeland, Florida 33802

Budget Size Bikini FREE
Director
U.S. Army Engineer Waterways Experiment Station
Post Office Box 631
Vicksburg, Mississippi 39181

Buffer Solutions Review 30 min., sd, b&w, 16mm, E.B.F. The pH scale of acidity is reviewed. Typical buffer reactions are shown. Thermochemistry is introduced. A calorimeter is shown.

Careers In Oceanography FREE
Assistant for Public Affairs of your Naval Commandant:
6th Naval District – U.S. Naval Base
Charleston, South Carolina

Challenges of the Oceans 29 min., sd, color, 16mm, McGraw-Hill, 1960. A scientific study of the physical, chemical and biological aspects of the oceans.

Challenge of the Sea FREE
Assistant for Public Affairs of your Naval Dist.

The Community 11 min., sd, color, 16mm, E.B.F., 1962. Examines three different ecological communities – the western pine forest, the grassland, and intertidal community.

Corrosion FREE
The Glidden Paint Company
Union Commerce Building
925 Euclid Avenue
Cleveland, Ohio 44115

Corrosion in Action FREE
Rothbacker Incorporated
241 West 17th Street
New York, New York 10011

C.S. "Long Lines" FREE
Local Bell System
representative or
affiliated company

Deep Frontier FREE
Assistant for Public Affairs of your Naval Dist.

Earth Beneath the Sea McGraw-Hill Text-Films. 30 min., sd, color.

The Electric Eel 12 min., sd, color, 16mm, Moody Inst., 1954. Describes the appearance, internal structure, respiratory system, and locomotive versatility of the electric eel.

Eyes in Outer Space 26 min., sd, color, 16mm, Disney, 1959. Describes work of weather stations: explores possibility of using satellites in long range forecasting, etc.

Fish Out of Water 11 min., sd, color, 16mm, Moody Inst. 1954. Animation and live action shots are used in showing how the spawning and hatching processes of the grunion are related to tide movements.

Gulf of Mexico Shrimp Trawls FREE
Gear Research Station
Bureau of Commercial Fisheries
P.O.Box 1909
Panama City, Florida 32402

History Layer by Layer McGraw-Hill Text-Films. 30 min., sd, color. Illustrates the significance of sediment layers with respect to the geologic history of the planet.

Identification of Sea Ice FREE
Assistant for Public Affairs of your Naval District
Request No. MN-7419-B

Inconstant Air 29 min., sd, color, 16mm, McGraw-Hill, 1960. Traces history of man's attempt to understand climate & weather. Explains how meteorological data is collected.

Indian Ocean Expedition FREE
Indian Information Services
2107 Massachusetts Ave. N. W.
Washington, D.C. 20008

Island Oddities FREE
Modern Talking Picture Service
3 East 54th Street
New York, N. Y. 10022

Introduction to Underwater Sound FREE
Assistant for Public Affairs of your Naval District *
Request No. MN-8857

Laboratory Techniques and Apparatus – I 30 min., sd, b&w, 16mm, E.B.F. Demonstrates laboratory techniques – how to light and regulate burners; how to cut and bend glass tubing; how to handle reagent bottles, etc.

Laboratory Techniques and Apparatus – II 30 min., sd, color, 16mm, E.B.F. Shows effective and safe ways of using laboratory apparatus: heating of liquids in test tubes, beakers and flasks; methods of evaporation, etc.

The Lemmings and Arctic Bird Life 21 min., sd, color, 16mm, Disney, 1964. The Lemming is a small rodent not usually found above ground before the snow is gone. With the spring thaw, Lemmings migrate to the sea: lakes and lagoons dot the landscape and become a haven for eider duck, turnstones, phalaropes, gulls and loons.

Life in the Sea Encyclopedia Britannica Films. 11 min., sd, color. Deals with elementary ecology; discusses plankton, free-swimmers, and bottom dwellers.

Making Solutions of Known Concentration 30 min., sd, b&w, 16mm, E.B.F. Two methods for expressing concentrations of solutions are discussed. Volumetric flasks are shown. Another method for preparing dilute solutions is shown.

Manager of the Sea FREE
Institute of Marine Science
University of Miami
Rickenbacker Causeway
Virginia Key, Miami, Florida 33149

Man Invades the Sea. McGraw-Hill Test-Films. 28 min., sd, b&w. Presents recent attempts and achievements of man in entering the underwater world.

Marine Borers FREE
Institute of Marine Science

Marine Corrosion FREE
Institute of Marine Science

Marshland Is Not Wasteland. Roy Wilcox Productions, Inc. 16 min., sd, color. Deals with the value of marshland as a natural resource.

Mission Oceanography FREE
Assistant for Public Affairs of your Naval Dist.

Mysteries of the Deep, 24 min., sd, color, 16mm, Disney, 1959. Explores the depths of the ocean to show some of the many interesting creatures to be found there.

Nature of the Sea Water, Du Art Film Laboratories. 30 min., sd, color. Describes the physical properties of temperature, salinity, and density.

Noisy Underwater World of the Weddell Seal. Sterling Educational Films. 11 min., sd, color. On an expedition into the Antarctic scientists go underwater to record the sounds seals make.

The Nuclear Ship "Savannah" FREE
Audio Visual Branch
Division of Public Information
U.S. Atomic Energy Commission
Washington, D.C. 20545

1,000 Feet Deep for Science FREE
Motion Picture Center
Westinghouse Electric Corp.
Gateway Center
Pittsburgh, Pennsylvania 15222

Ocean Currents 16 ½ min., sd, color, 16mm, McGraw-Hill, 1964. The ocean water is in constant motion, driven by tides, winds and currents. Vast quantities of water are moved from place to place on the earth's surface.

Oceanographic Studies Off George's Bank FREE
ESSA
Washington Science Center
Rockville, Maryland 20852

Oceanography; Science for Survival FREE
Assistant for Public Affairs of your Naval Dist.

Oceanography at Work (Diamonds under the Sea) 25 min., sd, color, 16mm, Coronet, 1966. Shows how geologists of today use seismic surveys coordinated with land-based radar beacons to map the ocean floor.

Ocean Tides: Bay of Fundy, 14 min., sd, color, 16mm, E.B.F., 1956. Presents scenes of the Bay of Fundy at high and low tides; explains what causes tides and how tidal range is determined.

Plankton and the Open Sea, 19 min., sd, color, 16mm, E.B.F., 1962. Discusses the importance of the minute plankton organism to marine food chains.

Plankton Pastures of the Ocean. Encyclopedia Britannica Films. 10 min., sd, color. Emphasizes the importance of plankton as the base of the food pyramid in the sea. Youngsters learn to recognize diatoms and dinoflagellates.

Plowshare FREE
Audio Visual Branch
Division of Public Information
U.S. Atomic Energy Commission
Washington, D.C. 20545

Problem at Port Washington FREE
U.S. Army Engineer Waterways Experiment Station
Corps of Engineers
Office of the Director
Vicksburg, Mississippi 39181

Project Mohole Report Number One FREE
The American Petroleum Institute
Committee on Public Affairs
1271 Avenue of the Americas
New York, N. Y. 10020

Protection of Mooring Buoys FREE
National Association of Corrosion Engineers
980 M & M Building
1 Main Street
Houston, Texas 77002

Protozoa 11 min., sd, color, 16mm, E.B.F., 1960. Dr. Roman Vishniac photographs an intimate close-up exploration of the world of one-celled animals.

Protozoa One-celled Animals 11 min., sd, color, 16mm, E.B.F., 1957. Dr. Roman Vishniac, identifies pseudopods, flagellates and ciliates, showing how they move, eat and reproduce.

Prowlers of the Everglades 32 min, sd, color 16mm, Disney, 1952. A photographic study of the wildlife of the Florida Everglades.

Putting Animals in Groups International Film Bureau. 13 min., sd, color. Introduces children to the idea that they can classify animals by observing their structures.

The Restless Sea FREE
Local Bell System
representative or
affiliated company

Return to Bikini FREE
Audio Visual Branch
Division of Public Information
U.S. Atomic Energy Commission
Washington, D.C. 20545

Science of the Sea. International Film Bureau. 19 min., sd, color. Demonstrates importance of the water cycle and of the sea-atmosphere relationship in weather prediction. Show collection and analysis of water samples and the application of the results to the study of water mass distribution.

The Sea 27 min., sd, color, 16mm, E.B.F., 1962. Depicts the interrelationships between living things in the sea, their dependence on each other and the varying conditions of the marine environment.

Sealab I FREE
Assistant for Public Affiars of your Naval Dist.
Request No. MN-10100

Secrets of the Underwater World 16 min., sd, color, 16mm, Walt Disney, 1956. Describes many of the unusual creatures of the shallow seas in the tidal fringe and in fresh water.

Seashore Life. Encyclopedia Britannica Films. 11 min., sd, color. Treats the seashore inhabitants of the East Coast and their mode of life. Provides a lesson in ecology through the portrayal of life on three kinds of seashores – the sandy beach, the rock pool, and the mud flat.

Sea Surface Meteorology. Modern Learning Aids. 24 min., sd, b&w. Explains by means of laboratory experiments, how salt particles act as nuclei for the formation of raindrops.

Ship Explorer Oceanographic FREE
Cruise 1960
ESSA
Washington Science Center
Rockville, Maryland 20852

The Shrimp FREE
Institute of Marine Science
University of Miami
Rickenbacker Causeway
Virginia Key, Miami,
Florida 33149

Sounds in the Sea 16 min., sd, color, 16mm, Moody Inst., 1955. Relates events during World War II leading to the discovery that fish produce sounds. Describes an undersea expedition.

Sponges and Coelenterates: Porous and Sac-like Animals 11 min., sd, color, 16mm, Coronet, 1962. Shows the cell specialization, sexual and asexual reproduction, and life cycles of sponges and coelenterates.

The Story of SeaLab II FREE
Assistant for Public Affairs of your Naval Dist.
Request No. MN-10100B

Survival in the Sea: The Life Cycle. Audio-Visual Center. 29 min., sd, color or b&w. Starts with the basic principles of plant growth. Shows the roles played by organisms in the food chain.

Terressrial Isotopic Power Systems FREE
Audio Visual Branch
Division of Public Informaiton
U.S. Atomic Energy Commission
Washington, D.C. 20545

Tidal Power FREE
Division Engineer
U.S. Army Engineer Division New England
424 Trapello Road
Waltham, Massachusetts 02154

Tides and Currents FREE
ESSA
Washington Science Center
Rockville, Maryland 20852

Time Lapse Study of Antarctic Ice Floes and Tidal Currents
Assistant for Public Affairs of your Naval Dist.
Request No. MN-10152

Tsunami FREE
ESSA
Washington Science Center
Rockville, Maryland 20852

A Typical Acid-Base Reaction: Indicators, 30 min., sd, color, 16mm, E.B.F. The Bronsted-Lowry theory is reviewed. New terms are introduced. Indicators are used to show how a solution is neutralized.

Voice Beneath the Sea Local Bell System Representative or affiliated company. FREE

Volcano Surtsey Capitol Film Laboratories, Inc. 30 min., sd, color. A record of the scientific expedition to study the birth and life of the volcano, Surtsey, which appeared recently in the North Atlantic.

Water Birds 30 min., sd, color, 16mm, Disney, 1952. A photographic study of a variety of water birds. Explains how nature adapted them to meet the problems of survival.

Water Masses of the Oceans. Du Art Film Laboratories. 45 min., sd, color, Deals with tidal currents and tidal circulations. Also touches on the explains by means of laboratory experiments.

Waves on Water 20 min., sd, color, 16mm, E.B.F., 1965. This film explores the manner in which water in a wave really moves. Explains how waves are created, describes wave refraction and discloses startling discoveries regarding hi-energy waves not produced by the wind.

What is a Fish? 22 min., sd, color, 16mm, E.B.F., 1963. Illustrates major types of fishes; examines anatomy of a typical fish; and demonstrates behavior patterns.

What is Ecology? 11 min., sd, color, 16mm, E.B.F., 1962. Explores the major biomes of the world from high arctic to the tropical rain forest.

Wood Preservation Effect on Marine Organisms FREE
Assistant for Public Affairs of your Naval Dist.
Request No. 8167-C

World of the Little Things 14 min., sd, color, 16mm, Moody Inst., 1954. Traces the development of the microscope. Uses timelapse photography and cinephotomicrography to show the structure and life processes of microscopic plants.